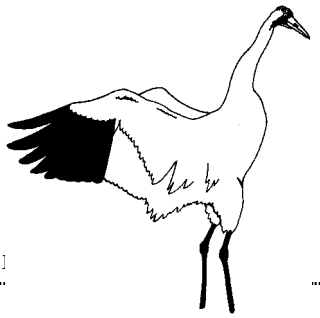
CHAPTER 8

Medicine and Surgery



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Success in captive rearing and propagation of cranes is dependent on establishing appropriate health monitoring, disease prevention, and parasite control procedures. A knowledge of common diseases, surgical procedures, anesthesia, and clinical pathology is crucial.

Initial Examination

History

Before the crane is removed from its pen or enclosure, the veterinarian should obtain a complete history. Questions asked should range from general information such as age, sex, and length of time in the collection, to those more specific, including:

1. Current clinical signs of disease including unusual behavior.
2. Medical treatment given to date.
3. History of clinical signs in the crane flock.
4. Recent changes in management procedures or in the crane's environment.
5. Unusual behavior (see Chapter 6): listlessness, reluctance to fly or move wings, lack of balance, restlessness, lameness or limping, straining, ruffled feathers, head held low, shivering, fainting, eyes partially closed, regurgitation, reluctance to rise, etc.
6. Changes in food and water consumption.
7. Appearance and consistency of droppings.
8. Overt signs of trauma or swelling in extremities.
9. Bleeding.

Physical Examination

The physical examination should be thorough but brief (i.e., normally not exceeding 10 min). A checklist (Fig. 10.5) and an assistant help assure thoroughness. Even when an obvious injury is present, a complete examination should be performed to detect less obvious problems or complications. A crane in critical condition should, of course, receive immediate treatment and be given a thorough examination later.

Head and Eyes. Restrain the crane (as described in Chapter 2). Examine the eye area for swollen lids, squinting, discharge, or a change in color of the globe. These changes can be due to injury, infection, foreign bodies under the lids, or swollen sinuses. Dilated pupils may indicate shock, concussion, or blindness. Hemorrhage in the anterior chamber or ear canal can be due to trauma to the head. A small light source is used to check pupillary response. In birds, pupils respond independently (i.e., no consensual reflex as in most mammals).

If abnormalities are noted or suspected during the initial superficial eye examination, the deeper structures of the eye should be examined using an ophthalmoscope. Because birds have striated rather than smooth muscle in the iris and ciliary body, atropine will not cause pupil dilation as in the mammalian eye. Iris color in Sandhill Cranes is blue in young chicks, changing to gray or gray-green, and finally yellow or orange as the bird matures. The retina is normally avascular. A dark, ribbon-like structure called the pecten is attached to the retina. The pecten is highly vascular and is considered to be the source of oxygen and nutrition for the retina.

Beak. The beak should be examined for bite and overgrowth. The sides of the beak should be checked for evenness of wear. Crane beaks grow several centimeters per year. As a result, some deformed beaks require frequent trimming (every 2-4 months). Beak trimming may also be required for non-deformed beaks during the winter when normal probing behavior is inhibited by frozen ground. When trauma is suspected, the beak should be palpated for fracture or other damage. The nares should be examined for any discharge or plugs.

Mouth. Often during the examination, the crane will open its mouth to vocalize, allowing a view of the structures inside. If not, the mouth can be opened by gently prying at the commissure with the index finger on one side and the thumb on the other side.

Normally, the mucous membranes are bright pink, but some cranes have gray or black pigmented tissues on the mucous membranes. Level of hydration can be estimated based on moistness of the mucous membranes. The tongue should be long and thin. There is a small, bright red structure at the tracheal opening (glottis) in Sandhill and Whooping Cranes.

Vitamin A deficiency can appear as proliferative, plaque-like lesions of the epithelium of the alimentary mucosa, conjunctiva, eyelids, ear canal, or skin. Protozoan (*Trichomonas*) and fungal (*Candida*) infections will occur as thick white, raised, plaque-like lesions covering the mucosa within the oral cavity and which may extend into the esophagus, proventriculus, etc. *Candida* is also seen as a lesion causing beak erosion. By contrast, scab-like lesions around the commissure of the mouth or on the eyelids are characteristic of the dry form of avian pox. "Wet" pox produces raised plaques in the oral cavity, esophagus, etc. and is considered a more severe, life threatening form of pox disease.

AUDITORY CANAL. Theauditory canals(Fig. 8.1) shouldbeexaminedforexudate,infection,orblood. In casesofsuspectedtraumaduetoaggression,thecanalis oftenswollen, partiallyclosed,and filledwith blood.

NECK. Carefully palpate the neck, trachea, and esophagus for the presence of solids, liquids, or gas (air). Because the lower part of the cervical esophagus of cranes, unlike many other bird groups, is not well developed as a storage area (crop), food and liquids generally pass quickly to the proventriculus, with the result that, on most examinations, the esophagus is empty. Gross distention can indicate blockage or impaction.

Deformities of the vertebrae (Fig. 8.2) are unusual in cranes. Scoliosis and wryneck are seen occasionally in young chicks (Fig. 8.3). On palpation, the neck cannot be extended straight, or will not assume a normal curvature.

THORAX. The thoracic body cavity should be examined by palpation and by listening to the heart, lungs, and air sacs using a stethoscope. A bilateral enlargement, found on palpation of the thoracic inlet, is associated with enlarged thyroids (goiter) as seen

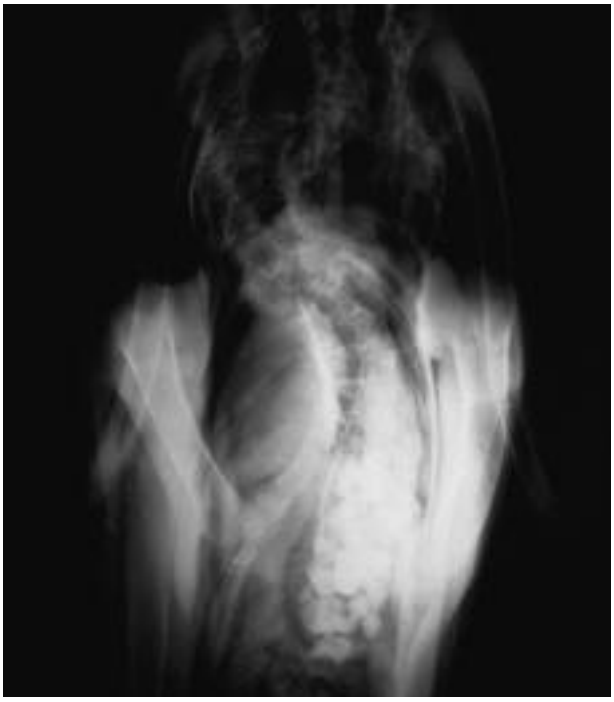

FIG. 8.2. *Vertebral deformities (radiograph).*
PHOTO GLENN H. OLSEN

FIG. 8.1. *Auricular feathers cover the auditory canal.*
PHOTO DAVID H. ELLIS

FIG. 8.3. *Wryneck in a Sandhill Crane chick.*
PHOTO GLENN H. OLSEN

in some bird groups, but not yet reported in cranes. Some cranes occasionally develop subcutaneous emphysema. In this condition, air-filled pockets under the skin occur along the thorax or over the thighs, abdomen, neck, and even the head (Fig. 8.4). Generally, trauma, with rupture of an air sac and leakage of air under the skin, is suspected as the cause, but often no wound can be found.

Fig. 8.4. *Subcutaneous emphysema over the head of a male Greater Sandhill Crane.* Photo David H. Ellis

Body Condition Index and Weight. By palpating the breast (pectoral) muscles and sternum (keel), the degree of development or atrophy of skeletal muscles can be estimated and is reported as the bird's body condition index (BCI) (Fig. 8.5). A BCI of 4 or 5 (on a scale of 1-5) is indicative of a well-muscled or plump bird and the pectoral muscles will be rounded convexly from the keel. A bird with a BCI of 3 will have a rather flat profile to the pectoral muscles. A BCI of 2 is a bird with a concave shape to the pectoral musculature, and a BCI of 1 indicates severe muscle atrophy and emaciation. Differences exist seasonally in individuals and from bird to bird: healthy wild cranes and captive cranes that fly free usually have more developed pectoral muscles than birds with flight impairment. Generally, birds with amputations of a wing have a loss of pectoral musculature, especially on the side of the amputation. The greatest value in using BCI is not the comparison between birds, but rather a comparison with previous BCI readings for the individual. Caretakers should be trained to palpate and record BCI any time a crane is handled.

A drop in BCI normally indicates weight loss and a possible medical problem. Weighing is the best method for evaluating body condition and monitoring a bird's overall nutritional status. Direct weights can be taken as described in Chapter 2 (see Fig. 2.9). Weight loss signals the need for a complete examination by the veterinarian.

Abdomen. Gently palpate the abdomen for internal masses, fluid (ascites), or ovulated eggs. Generally the liver is not palpated unless enlarged. The gizzard and intestines are easily palpated, and any crepitus (gas), excess fluid, thickening, or masses in the intestines are possible to detect. The vent area should be examined for lesions, growths, protrusions, and for feces or urates accumulating on the feathers. A soiled vent in young chicks is frequently a sign of diarrhea, often caused by *Escherichia coli* infections (see Chapter 5, Veterinary Techniques section). The uropygial or preen gland above the base of the tail can be palpated for enlargement which can be caused by neoplasia, impaction, or infection.

Skin and Plumage. Examine the general condition of the skin and plumage. Look at the general level of hydration as indicated by the elasticity of the skin. Look for mites, lice, skin swellings (emphysema, abscesses), and missing or damaged feathers. Dull, split, or frayed feathers and stress bars across the feathers can indicate nutritional deficiencies, hormonal imbalances, or stress. Feather cysts and abnormally developed feathers are sometimes seen, especially on the wings. Skin irritation and broken feathers or an area of missing feathers on the thighs are seen in

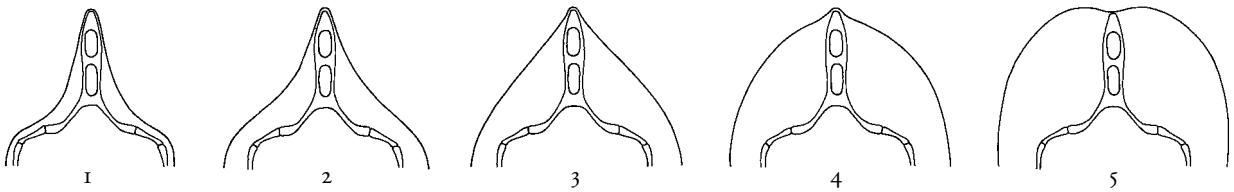

Fig. 8.5. *Body Condition Index (BCI) is an indicator of nutrition: cross section through mid sternum of Sandhill Crane (cavities are tracheal passages).* Art Kate Spencer

self-mutilation. A similar condition is often associated with reproductive activity in male cranes in the spring especially after being handled for artificial insemination (AI) for an extended period.

WINGS. Examine each wing, palpating all bones and joints while also assessing muscle tone and extension. Be careful to maintain the wing in a natural position to avoid injury. Check the wings for swelling, bruising, or abrasions which are particularly common on the point of the carpus (wrist area). Subcutaneous hemorrhage in cranes will develop a green discoloration due to biliverdin (green pigment) released as red blood cells (RBCs) are destroyed. Matted feathers on a wing or over the body often indicate a wound.

LEGS. Palpate each leg bone and joint while assessing muscle tone. Check the toes for normal extension, for broken nails, and for swollen areas on the toes or plantar foot. Toe swellings occur with fractures, luxations, and bumblefoot (pododermatitis), an infection frequently caused by *Staphylococcus aureus*. Cranes with a leg injury that forces them to stand on one foot, often develop pressure necrosis of this foot with resulting open wounds that develop into bumblefoot.

AUSCULTATION. Auscultation of the chest should be included in every examination. Using a stethoscope, it is possible to determine heart rate, rhythm, and location of sound as well as to detect cardiac murmurs in chicks as young as 3 days. Some of these murmurs resolve within the first few days after hatching. Others are due to the most common congenital anatomic anomalies (i.e., atrial or ventricular septal defects). Other murmurs have been detected in severely anemic or dehydrated cranes and in cranes with pericarditis due to infection or visceral gout. Arrhythmia and cardiac tamponade have also been detected in cranes. Auscultation of the thorax is also useful for assessing the respiratory system. Unlike smaller birds, crane respiration produces distinct sounds associated with air movement. These sounds are normally louder on inspiration than expiration.

Clicks, wheezes, fluid-type sounds, or total absence of air movement sounds in one or more location can indicate a respiratory problem. Occasionally, unusual respiratory sounds are found on one examination and not on the next. The cause of these transitory sounds are unknown, but apparently are not related to major disease problems. Unilateral, dull respiratory sounds can indicate blockage of the major bronchi, or consolidation of lungs and air sacs on one side, and are often heard in advanced cases of aspergillosis.

TEMPERATURE. Unlike smaller birds, cranes appear to have a relatively constant body temperature (40.7° C, +0.4° [105.3° F, +0.8°] in Whooping Cranes and 40.5° C, +0.6° [104.7° F, +1.2°] in Sandhill Cranes). Although elevated temperatures have been observed in cranes suffering from bacterial disease, exertion, and stress, cloacal temperature monitoring is not commonly part of the physical examination.

Initial Care of a Sick Crane

After a sick or injured crane is found, it may be some time before a clear diagnosis is made. Causes for not having an immediate definitive diagnosis can be: (1) the crane is too weak to undergo extensive testing, (2) tests being performed for the suspected condition take time (sometimes days or weeks) for results to be received, (3) the crane's condition is attributed to multiple etiologies, not all of which have been properly identified, and (4) the condition is difficult to diagnose because the initial cause is no longer present. Having collected the required samples to confirm the clinical diagnosis, the practitioner must begin therapy until the results from the tests arrive.

Even prior to diagnosis, it is often important to begin some form of therapy. Of all the initial therapies, fluid therapy is the most important except in cases of severe anemia or hypoproteinemia. For cranes in shock from disease or trauma, bolus intravenous therapy has been useful (Redig 1984). We recommend using lactated Ringer's solution because of its similarity to avian plasma. Normal saline is also a good choice. Estimate the dehydration level (10% dehydration is a suitable estimate if dehydration is detected). The maintenance fluid requirement for a crane is about 44 mL/kg/day. Approximately 50% of the deficit should be given in the first 12 hours. The remainder of the deficit and the calculated maintenance dose should be given over the next two days. The fluids should be kept warm (35.5° C, 96° F) in an incubator or warm water bath.

Fluids can be delivered by one of several routes. The initial calculated dose of fluid can be administered as a bolus intravenously. The jugular or the medial tarsal vein are the easiest veins to access in the crane patient, although the ulnar or brachial vein can be used. Care needs to be taken to ensure that there is no further blood loss from hematoma formation, especially when using the jugular or brachial vein for

this procedure. Apply pressure over the venipuncture site for 1-2 min immediately after withdrawal of the needle. Using a small-gauge needle (25-gauge [5/8-in] or 26-gauge [3/4-in]) or butterfly needle may also help. For repeated intravenous therapy, a catheter (20-23-gauge in adults) can be placed in the medial tarsal, jugular, or brachial vein (under anesthesia).

Another equivalent route for frequent fluid administration is the intraosseous route (Ritchie et al. 1991). For this technique, feathers are removed from the carpal joint area, and the site is scrubbed with a disinfectant soak, rinsed, and sprayed with alcohol or iodine. A long needle (16-20-gauge, spinal needle) is used as a drill to pierce the distal cortex of the ulna and enter the marrow cavity. You should then be able to aspirate marrow. Flush the catheter with saline or heparin saline, then place an injection cap over the end of the needle. Suture or glue the needle to the skin of the wing. Finally, the wing is bandaged in a Figure-8 pattern to restrict movement and to cover the exposed needle hub and cap, protecting it from the probing beak of the crane.

In non-critical cases, fluids can be administered by the subcutaneous route. The advantage of this route is that it is the least traumatic for the crane and can easily be used for repeated dosages. The disadvantage is that absorption rates are slower, often as long as 30 min in a healthy crane and longer in severely depressed cranes. The best areas for subcutaneous fluid administration in cranes are the flanks just in front of the legs, the base of the neck, or intrascapular area.

With dehydration and capture myopathy, there may be an accompanying alteration in the acid-base balance resulting in acidosis. Ideally, the level of acidosis should be determined through laboratory testing, but immediate results are not always available. Therefore, if acidosis is suspected, a crane can be given 1 mEq/kg of sodium bicarbonate subcutaneously with fluids every 30 min to a maximum of 4 mEq/kg. Fluids containing dextrose are considered acidifying agents and should be used carefully in the dehydrated or acidotic crane. It is rare for a sick adult crane to be severely hypoglycemic. However, for young chicks, giving glucose or dextrose orally or by injection may be critical to survival.

For sick birds in general, a heated environment (30-32° C, 85-90° F) is helpful. This is especially true in very young crane chicks. Often sick older cranes are brought into a room heated to 21° C (70° F) and equipped with a heat lamp to raise the temperature in one area up to about 30° C (85° F). A recumbent crane should be placed under the heat lamp, but continuously monitored for signs of overheating. When the crane becomes more active, it will move in or out from under the lamp as it requires heat supplementation.

Several medications may be indicated when the sick or injured crane is first presented. Corticosteroids such as dexamethasone or methyl prednisolone (see Table 8.1 for dosages) are indicated in cranes suffering from acute trauma or shock. Antibiotics are required in some traumatized and sick cranes. Ideally, the antibiotic chosen is based on cultures of the disease site and sensitivity testing, but results from such testing can take 4-7 days. Therefore, the clinician must decide on initial antibiotic therapy based on previous experience with the disease and the expected types of bacteria to be isolated by culturing.

Enrofloxacin, ampicillin, an aminoglycoside-penicillin combination such as amikacin and piperacillin sodium (with supplemental fluid therapy), and trimethoprim-sulfa combinations are each used on certain cases (see Table 8.1 for dosages). Enrofloxacin is a useful broad spectrum antibiotic, however, in young mammals, this drug has been shown to interfere with joint cartilage development. Although ontogenic problems have not been reported in any bird, use this drug with caution. Ampicillin is used with the injured, dehydrated crane because of this drug's safety, low renal toxicity, and bacteriocidal properties. Aminoglycoside-penicillin combinations are useful in sick cranes, especially for respiratory diseases, but not in cases involving impaired kidney function because aminoglycosides can be toxic to the kidneys.

Vitamin injections can be given as part of the supportive program (see dosages in Table 8.1). Vitamin A is important in cases where a deficiency is suspected (see initial examination) or in bumblefoot. Iron dextran injections (see Table 8.1 for dosage) are given in cases of anemia as indicated by a low hematocrit and pale mucous membranes. The use of iron dextran has been shown useful in raptors (Redig 1984), but no similar tests or dramatic results have been seen in cranes.

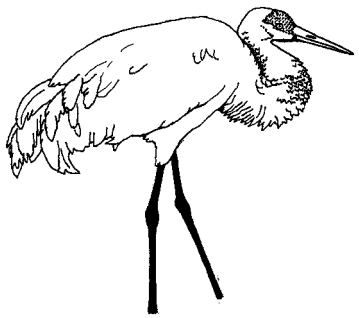

TABLE 8.1

Medications commonly used for cranes.[1]

DRUG	INDICATIONS	ROUTE OF ADMINISTRATION[2]	DOSAGE	TREATMENT SCHEDULE
ANTIBIOTICS				
Amikacin	Broad-spectrum, less nephrotoxic than gentamicin; often used in conjunction with piperacillin sodium; ensure adequate hydration	IM	10 mg/kg	2/day
Ampicillin	Broad-spectrum antibacterial drug for gram-negative and gram-positive bacteria, useful for several pathogenic enteric organisms	IM	100 mg/kg	2/day
Carbenicillin	Good only for 3 days after mixing; synergistic with aminoglycosides	IM,IV	100 mg/kg	2-3/day
Cefotaxime sodium	Broad-spectrum; sometimes used in conjunction with aminoglycosides	IM	50-100 mg/kg	3/day
Cephalexin	Broad-spectrum; effective against most gram-positive organisms and some gram-negative organisms, including various enteric organisms	oral	35-50 mg/kg	4/day
Cephalothin	Same as cephalexin	IM	100 mg/kg	4/day
Chloramphenicol	Broad-spectrum activity against both gram-positive and gram-negative bacteria, rickettsia, and chlamydia	SQ	100 mg/kg	3/day
Enrofloxacin	Broad-spectrum antibiotic	IM, oral	8-15 mg/kg	2/day
Gentamicin	Broad-spectrum; used therapeutically to treat bacterial infections in cranes and prophylactically against bacterial infections in newly hatched chicks; ensure adequate hydration	IM	5 mg/kg	2-3/day (1 in newly hatched chicks)
Piperacillin sodium	Used with amikacin	IM	100 mg/kg	2/day
Trimethoprim sulfa	Respiratory and enteric infections, also used as anticoccidial; regurgitation common orally	oral IM	16-24 mg/kg[3] 8 mg/kg[4]	2-3/day 2/day
Tylosin	Effective against gram-positive and some gram-negative bacteria mycoplasma, and chlamydia; useful for respiratory infections	SQ	15 mg/kg	3/day
CORTICOSTEROIDS				
Dexamethasone	Shock, trauma, endotoxemia capture myopathy, etc.	IM, IV, SQ	2-8 mg/kg (reduce doses for long-term therapy)	1-2/day
Prednisolone	Shock, trauma, chronic lameness	IM, IV	2 mg/kg	1-2/day
Prednisolone sodium succinate	Shock	IM, IV	10-20 mg/kg	as needed

TABLE 8.1 CONTINUED

Medications commonly used for cranes.

Drug	Indications	Route of Administration[2]	Dosage	Treatment Schedule
Vitamins				
Vitamin A (Aquasol A) 100,000 units/mL	Hypovitaminosis A, sinusitis, ophthalmic diseases, avian pox	IM	1.0 mL/kg	twice weekly
Vitamins A, D_3, E (Injacom-100) 100,000 units A, 100,000 units D_3/mL	Hypovitaminosis A, ophthalmic diseases, fractures, egg binding, soft shelled eggs, and respiratory infections (especially sinusitis)	IM	1.0 mL/kg	once weekly
Vitamin B complex	CNS signs, trauma, muscular weakness, anemia, debilitation, and anorexia	IM	1-3 mg/kg	1/day
Vitamin E and selenium (50 mg Vitamin E, 1 mg Se/mL)	Muscular weakness, capture myopathy, leg dysfunctions, prior to or at times of capture or stressful event	IM	0.05-0.10 mg/kg	once every 14 days
Injectable Tranquilizers and Anesthetics				
Ketamine HCl	Disassociative anesthetic	IM	10-22 mg/kg	once, lasts 10-30 min
Xylazine	Tranquilizer, given with ketamine	IM	1.0-2.2 mg/kg	once
Diazepam	Tranquilizer, use alone or with ketamine	IM	0.5-1.0 mg/kg	once, lasts 2-6 hr
Midazolam	Tranquilizer	IM	15 mg/kg	once
Yohimbine	To reverse xylazine	IV	0.1 mg/kg	repeat in 10 min as needed
Tolazoline	To reverse xylazine	IV	15 mg/kg	once
Flumazenil	To reverse midazolam	IM	0.1 mg/kg	once
Other Medications				
Doxapram	Respiratory stimulant	IM, IV	5-10 mg/kg	1/day
Iron dextran	Following hemorrhage or for iron deficiency anemia	IM	10 mg/kg	once every 7-10 days if hematocrit is still low
Phenylbutazone	Non-steroid anti-inflammatory, and anti-pyretic	Oral	3.5-7 mg/kg	2-3 times per day
Methocarbamol	Muscle relaxant	Oral IV	50 mg/kg 32.5 mg/kg	once

[1] *From Custer et al. 1979; Bush et al. 1979, 1981a, 1981b; Lock et al. 1982; Carpenter 1986, 1993; Klein et al. 1994; Olsen and Carpenter 1996.*

[2] *IM = intramuscular; SQ = subcutaneous; IV = intravenous.*

[3] *Dose based on trimethoprim suspension (8 mg trimethoprim and 40 mg sulfamethoazole/mL).*

[4] *Dose based on trimethoprim 24% for injection (4 mg trimethoprim and 200 mg sulfadiazine/mL).*

Nutritional Support of a Sick Crane

If a crane is not eating, supplemental feeding (i.e., tube feeding or gavage) may be required. Insufficient intake of calories can lead to emaciation quickly in a sick crane. The bird will first mobilize body fat and then will catabolize muscle. Severe nutritional depletion is indicated by a drop in blood glucose; most normal adult cranes will maintain a blood glucose over 180 mg/100 mL (Table 8.2A). Restoring and maintaining a positive energy balance and adequate protein, vitamin, and mineral intake is critical in the treatment of sick cranes.

Tube feeding of cranes is easily accomplished with one person holding the crane in the normal fashion (Fig. 5.16), while a second person grasps the head and, using the thumb and fore-finger, gently forces the beak open. With the other hand, a flexible tube (we use red rubber urinary catheters, size 10-16 French) is inserted into the mouth, over the tongue, and down the esophagus to the level of the thoracic inlet or proventriculus. Care must be taken at this point to determine that the tube is in the esophagus and not the trachea. This is done by inspecting the mouth and seeing that the tube passes over the glottis. When the tube is in place properly, two cylindrical structures can be palpated, the trachea and the tube in the esophagus. If the tube is in the trachea, then only one cylindrical structure will be felt, and the tube should be removed immediately and reinserted properly before administering the food.

A 30-140-cc syringe containing the chosen tube feeding formula (see below) is attached to the tube, and the food is gently forced down the tube while watching the mouth for regurgitation. If the bird starts to regurgitate, stop tube feeding. First **remove the tube**, then quickly clean out the mouth especially around the airway, and then gently stroke the neck in a downward motion to encourage swallowing. Decrease the amount of formula fed on subsequent tubings to prevent further regurgitation. Because tube feeding also contributes to fluid balance, adjust total fluid therapy accordingly.

In the severely emaciated crane, an initial tube feeding or two should be of a high carbohydrate, low protein diet. Products useful in such situations are Emeraid I and some human enteral diets. Emeraid I is usually mixed 1:1 with water, though more dilute preparations can be used. After several successful feedings and normal fecal production, a more complex diet providing protein, fat, and fiber may be tube fed long term. Several different formulas have been used successfully in cranes. One common approach is to make a gruel from the crane's pelleted diet then supplementing with several additional nutrients (e.g., Table 5.3). Other choices are Emeraid II (mixed 1:1 with warm water) and liquid human enteral products.

Most of these formulations have a caloric density of 0.5-1.5 kcal/mL. To determine the amount of tube feeding formula that the crane needs each day, calculate the adult crane's daily energy requirement (see also the Nutritional Support section of Chapter 5) using this formula:

Daily Energy Requirement (Kcal) = 1.5 × Basic Metabolic Rate (BMR).

$BMR = 78 \times Weight_{kg}^{0.75}$ (Quesenberry et al. 1989).

The daily energy requirement divided by Kcal/mL of the formula equals mL of formula needed per day. The total volume should be divided into 2-4 meal. Do not feed more than 150 mL at one meal. It is best to start with meals of 60-80 mL.

Once a crane is eating normally again, tube feeding can be discontinued. The crane needs to eat approximately 100-200 g of pelleted food daily to maintain its body weight. It is often better to allow a crane some weight loss as it feeds itself rather than to continue the stressful practice of tube feeding.

Clinical Pathology

Hematology

Blood parameters are invaluable in diagnosing diseases and monitoring health. It is advisable to establish baseline blood profiles for your collection. Standard avian hematology techniques are appropriate for cranes. Excellent hematology references, complete with color plates of avian blood cells, are Dein (1984), Campbell (1988, 1994), and Hawkey and Dennet (1989).

EQUIPMENT FOR BLOOD COLLECTION. Blood can be collected using a 1-cc, 3-cc, 10-cc, or 60-cc syringe. The only use for the 60-cc syringe is collecting blood for transfusions. A 25-gauge needle is adequate for collecting small amounts of blood, but larger needles (20-23-gauge) or a catheter are preferred for collecting larger samples.

TABLE 8.2A

Normal hematologic and serum chemistry values for captive cranes[1]

	Sandhill Crane		Whooping Crane		Siberian Crane		Red-crowned Crane		Wattled Crane	
	Mean	Range	Mean	Range	Mean	Range	Mean	Range	Mean	Range
Hematocrit (%)	43.0	37-49	42.0	38-48	45.0	40-50	39.0	33-45	44.8	40-50
Hemoglobin (g/100mL)	13.5	10.5-18.7	14.4	13.0-16.7	—	—	—	—	—	—
Red Blood Cell (10^6/mm^3)	2.5	1.9-3.3	2.2	1.8-2.6	—	—	—	—	—	—
White Blood Cell (10^3/mm^3)	13.0	6.2-22.6	18.2	12.2-25.1	10.8	6.5-15.0	14.9	6.3-23.5	12.7	3.2-22.2
Total Protein (g/100mL)	3.9	2.9-7.9	3.8	3.1-4.4	3.6	3.1-4.1	3.3	2.9-3.6	3.1	2.8-3.4
Albumin (g/100mL)	1.5	1.0-2.5	1.5	1.2-1.7	1.4	1.2-1.5	1.2	1.1-1.3	1.1	1.0-1.3
Globulin (g/100mL)	2.3	1.8-3.4	2.3	1.8-2.8	2.3	1.9-2.7	2.1	1.9-2.3	2.0	1.8-2.1
Albumin/Globulin	0.6	0.4-1.3	0.7	0.6-0.8	0.6	0.5-0.7	0.6	0.5-0.7	0.6	0.4-0.6
Alkaline Phosphatase (IU/L)	164.0	34-423	46.0	28-72	45.2	28-68	226.4	128-409	37.2	13-61
Lactic Dehydrogenase (IU/L)	278.0	108-488	440.0	178-975	202.3	100-323	287.5	161-372	137.0	55-249
Aspartate Aminotransferase (IU/L)	181.0	16-260	261.0	133-612	181.6	117-254	208	108-456	188.5	148-230
Alanine Aminotransferase (IU/L)	50.0	19-162	53.0	42-71	16.1	6-25	29.6	18-67	10.5	10-11
Glucose (mg/100mL)	247.0	87-323	232.0	210-267	266.4	209-314	267.4	239-328	266.2	246-293
Uric Acid (mg/100mL)	9.7	4.1-24.6	8.1	6.5-10.2	9.0	5.5-12.6	7.8	4.2-11.3	7.7	5.4-11.1
Creatinine (mg/100mL)	0.7	0.4-1.2	0.6	0.4-0.8	0.3	0.3-0.4	0.3	0.2-0.4	0.4	0.3-0.5
Cholesterol (mg/100mL)	128.0	87-187	148.0	96-200	212.3	148-286	170.2	140-217	147.0	120-188
Creatine kinase (U/L)	—	—	—	—	106.1	48-205	114.3	53-333	76.7	19-139
Triglyceride (mg/dL)	—	—	—	—	142.4	128-555	265.4	108-520	110.3	58-169
Iron (mg/dL)	—	—	—	—	106.3	61-155	100.5	61-150	161.3	109-238
Calcium (mg/100mL)	9.7	8.8-10.9	9.1	8.3-9.7	10.5	9.5-11.2	10.8	10.4-11.2	10.8	10.3-11.6
Phosphorus (mg/100mL)	3.6	1.7-5.4	2.8	2.0-4.1	3.8	1.9-5.8	3.5	2.0-4.4	2.7	1.6-5.7
Sodium (mEq/L)	148.0	142-160	147.0	140-152	148.5	146-151	147.5	143-150	146.3	144-153
Chloride (mEq/L)	108.0	101-115	107.0	102-113	109.1	106-113	107.0	104-110	108.3	105-111
Potassium (mEq/L)	3.4	2.2-4.8	3.4	2.6-4.2	2.9	1.6-4.0	2.8	1.6-4.0	3.2	2.0-3.9

[1] *Based on Gee et al. 1981; Carpenter 1986; and unpublished work at Patuxent and ICF.*

BLOOD COLLECTION. Sample requirements vary according to laboratory and test. The maximum blood volume that can be safely collected is 1% of the crane's body weight (1 cc/100 g). Some laboratories require blood to be collected into heparinized capillary tubes (see Appendix), others require heparin or ethylenediaminetetraacetic acid (EDTA) to be used in the test tube to prevent coagulation of the blood. Blood from African Crowned Cranes will sometimes "sludge" or begin to clot even when EDTA is used as an anticoagulant; therefore, heparin is preferred for these species.

There are **three major venipuncture sites** for cranes. The preferred site, especially for large samples, is the **right jugular vein**. The crane is held in a normal carrying position (Figs. 2.6 and 5.4) while a second person, standing to the left of the head, holds the head and neck outstretched and rotated so the right side of the neck is up. With the second hand, this person applies firm pressure at the base of the neck, causing the right jugular vein to fill with blood. Wetting the feathers with alcohol makes the vein more visible. The person doing the venipuncture then moves in from the right and collects the required blood sample by inserting the needle through the skin into the jugular vein (Fig. 8.6). Pressure should be applied to the jugular vein at the puncture site for a minimum of 1 min after the needle is withdrawn to prevent the development of a hematoma. If the crane struggles or moves during the venipuncture, the needle may lacerate the wall of the vein and produce a hematoma. On rare occasions, this has resulted in the death of a crane. If a hematoma results, continue to apply pressure over the jugular vein at the hematoma site until the hematoma is no longer enlarging. Observe the crane for the next 30 min after release for signs such as ruffled neck feathers, enlarged neck, lethargy, or collapse.

FIG. 8.6. *Venipuncture of jugular vein.* PHOTO GLENN H. OLSEN

The **medial metatarsal vein** is another preferred site for venipuncture. One person holds the bird in the normal carrying position and applies pressure to the extended leg above the hock joint. This causes the medial metatarsal vein to be prominent along the inside of the hock and tarsometatarsus. A second person cleans a small area with alcohol, grasps the leg below the venipuncture site with one hand, and with the other hand, draws the sample with the syringe. This technique works well in large cranes, but in smaller cranes, the vein is smaller and frequently collapses as blood is withdrawn. Only small amounts of blood (up to 5 cc) can be withdrawn from this vein, and the vein will collapse if blood is drawn quickly. Other disadvantages of this site are the difficulties in restraining the leg and the risk of injuring the leg if the bird struggles. Advantages are that hematomas are rare because minimal subcutaneous space is available for blood to pool between the bone, tendons, and scales. Also, if bleeding does occur, a pressure bandage can be easily and safely applied.

The third site commonly used for venipuncture is the **brachial vein** (cutaneous ulna vein) on the underside of the wing in the area of the elbow. This requires that the bird be held supine (breast up) on a flat surface (ground, table, etc.) with one wing extended. A second person applies pressure over the humerus to help fill the brachial vein. Clean the site with alcohol to separate the feathers and expose the vein. A third person moves in from the caudal aspect of the wing to obtain the sample. Up to 10 cc can be collected from this site. A disadvantage, though, is the difficulty of safely restraining the bird in this position and the increased risk of injury. Small hematomas often occur at this venipuncture site. This technique has been used to obtain blood samples from anesthetized cranes where manipulation of the neck may interfere with delivery of the gaseous anesthetic agent.

MAKING BLOOD SLIDES. The conventional method of using two slides to make a smear will damage avian cells unless a beveled edge slide is used to make the smear. Another technique is to dilute the blood 1:4 with 30% bovine albumin solution (Olsen and Gaunt 1985). A second method of making blood smears is to place a drop of blood on a glass coverslip, then place a second glass coverslip (or a slide) on the first, allowing the blood drop to spread between the coverslips. Thereafter, quickly slide the coverslips apart, leaving a smear of blood on each slip.

STAINING SMEARS. Smears can be stained with a Wright or Wright-Giemsa stain, using Cameo Quick

Stain II or Diff-Quick (see Appendix). The three solutions used in staining slides (Wright-Giemsa technique) are methanol, phosphate buffered eosin, and phosphate buffered thiazine. Slides are first fixed in methanol for 10-15 sec, then stained for 10-15 sec in each of the other two solutions, and finally rinsed in distilled water.

Evaluating Blood Smears. When reading blood smears made by the two-slide method it is important to read all areas of the slide (both the edge and center of the smear) to get an accurate differential cell count. When reading slides made by the cover glass method read only the central portion of the smear.

Crane **red blood cells** (RBCs) are oval or elliptical. Each cell has a central nucleus, shaped similar to the cell and consisting of dark purple clumps of chromatin. Cytoplasm is slightly orange-yellow to pink tinged. Immature RBCs have blue-tinged cytoplasm, a lighter nucleus (less dense chromatin), and may be more prevalent in cases of anemia.

Thrombocytes are similar in function to mammalian platelets. Their cytoplasm is pale blue and may contain 1-3 small magenta granules which help to differentiate thrombocytes from immature RBCs or lymphocytes. The thrombocyte nucleus is dense and dark purple when stained. Thrombocytes are often found in clumps.

Heterophils are round with clear to pink cytoplasm containing usually elongated, pink, red, or purple, granules. The nucleus is blue to purple and can have two or more lobes. The cytoplasm of heterophils is normally clear, but toxic heterophils have deep blue vacuolated cytoplasm.

Eosinophils are round with pale blue cytoplasm and bright red, round to oval, granules. The nucleus is blue to purple with two or more lobes. The primary distinguishing characteristic between heterophils and eosinophils is the shape of the granules (elongated in the heterophil, round in the eosinophil).

Basophils are also round but have clear cytoplasm with deep purple granules and a blue or purple nucleus. Basophils are extremely rare cells, about 1-2 cells per 500 white blood cells (WBCs) counted.

Monocytes are large and irregular in shape. They have a light blue cytoplasm that may be vacuolated. The nucleus is often eccentrically located, round or elongated, and bilobate.

There are two types of **lymphocytes** seen in avian blood. Type I lymphocytes are similar in shape and appearance to thrombocytes. They have a small amount of deep blue cytoplasm. The nucleus is large compared to cell size, blue to purple colored, round, and eccentrically located. Type II lymphocytes are larger than type I lymphocytes and can be confused with monocytes. They have pale blue cytoplasm, a blue irregular shaped nucleus and higher cytoplasm-nucleus ratio.

Hematocrit. The hematocrit (HCT) or packed-cell-volume (PCV) is measured by filling a microhematocrit tube and spinning the tube in a high speed centrifuge to obtain a centrifugal force of 2260G (ca 3800 rpm for 10 min in a No. 2 International Centrifuge [see Appendix] [Campbell 1988:7]). Read the results by measuring the height of the RBC column over the height of the total column (cells and serum) and express as a percentage. Light yellow coloring of the serum or plasma can be due to carotenoid pigments (Dein 1984) and does not necessarily indicate icterus (abnormal yellowing of serum and some tissues). Lipemia (elevated fat) may be present as a milky-white serum in obese birds, post-feeding birds, and laying females.

Hemoglobin. Hemoglobin (Hb) measurements are important because they relate to the ability of the RBCs to transport oxygen. Hemoglobin measurements can be made using standard methods developed for mammals.

One of the oldest methods uses a hemoglobin scale. A drop of whole blood is placed on white filter paper, and the resulting red dot is matched for intensity with the best choice on a red color chart. This method is rapid, simple, inexpensive, and a good technique under field conditions, but the error is ±10-40% (Schalm et al. 1975:52-55).

Another simple method, but one requiring the purchase of equipment, is the oxyhemoglobin method. In this procedure, a drop of blood is placed on a glass plate, and the cells are destroyed using a hemolytic agent (usually saponin) dried on the end of a special applicator stick. Then a second glass plate is placed over the first, and the two are pressed together. The glass plates are then placed in the Spencer hemoglobinometer (see Appendix). Finally, the green color of the sample is matched against the standards read through the hemoglobinometer. Green is the color used for matching because the maximum absorption of hemoglobin under visible light occurs in the green band, and for the human eye, green is an easy shade to match accurately (Coles 1980:79-83).

An extremely accurate technique for hemoglobin is the cyanmethemoglobin method. The first step is to prepare a 1:200 dilution of blood in 0.04% ammonium hydroxide. Then the solution is placed in a

spectrophotometer or a filter photometer and read at 540 mμ. The percent transmission at 540 mμ is compared to a standard solution of cyanmethemoglobin, converting percent transmission to grams of hemoglobin per dL of blood (Schalm et al. 1975:52-55; Coles 1980:79-83).

RED BLOOD CELL COUNT. The total RBC count can be obtained using a Coulter counter or by the Unopette system (see Appendix) using standard techniques as in mammals (Dein 1984).

Mean corpuscular volume (MCV):

MCV = (HCT/RBC) × 100

Mean corpuscular hemoglobin concentration (MCHC):

MCHC = (Hb/HCT) × 100

Mean corpuscular hemoglobin (MCH):

MCH = (Hb/RBC) × 100

The above calculations are also used for other species of birds and mammals, but the reference values for normal individuals of most species of cranes need to be established. Table 8.2A contains some normal values for Whooping, Sandhill, Siberian, Red-crowned, and Wattled Cranes.

WHITE BLOOD CELL (WBC) COUNT. Because avian RBCs are nucleated, the techniques used to separate red and white blood cells in mammals will not work. Therefore, automated counters such as the Coulter unit cannot be used. Similarly, hand-counting systems such as the Unopette WBC count will not work with crane blood. The Eosinophil Unopette method is the most useful technique for estimating WBCs in cranes. However, the Eosinophil-Unopette method counts only heterophils and eosinophils. Mononuclear cells (monocytes and lymphocytes) are not counted, but the count is corrected to include these cells based on the differential count (ratio of various WBCs seen on the blood smear). The procedures to follow are:

1. Use the protective cap over the Unopette pipette tip to puncture the diaphragm on the small reservoir containing the stain.
2. Fill the pipette until blood reaches the neck.
3. Carefully wipe the tip of the pipette.
4. Gently squeeze the reservoir containing stain as the pipette is inserted securely into the reservoir.
5. Mix the blood and fluid in the reservoir by inverting 10-15 times.
6. Wait 5-10 min for staining.
7. Switch the pipette direction so it can now be used to fill a hemacytometer. Gently invert 3-4 times to uniformly resuspend cells.
8. Discard the first 10 drops and then fill both sides of the hemacytometer.
9. Place the hemacytometer in a covered petri dish containing a small amount of moistened filter paper (to prevent loss of fluid due to evaporation from the hemacytometer). Leave the hemacytometer undisturbed for 5 min to allow the cells to settle. Read the sample within 20 min of beginning the test, because RBCs will also eventually absorb the stain.
10. Count purple-stained cells in all 9 bold squares of both sides of the hemacytometer (Fig. 8.7). Find the total heterophil-eosinophil count (H/E) by multiplying the total number of cells counted by 17.6. An alternative method is to count 5 bold squares on both sides of the hemacytometer (Fig. 8.7) and multiply by 32.
11. Evaluate the WBCs on a stained blood smear for a differential count (Table 8.2B: usually based on counting 100-200 WBC).

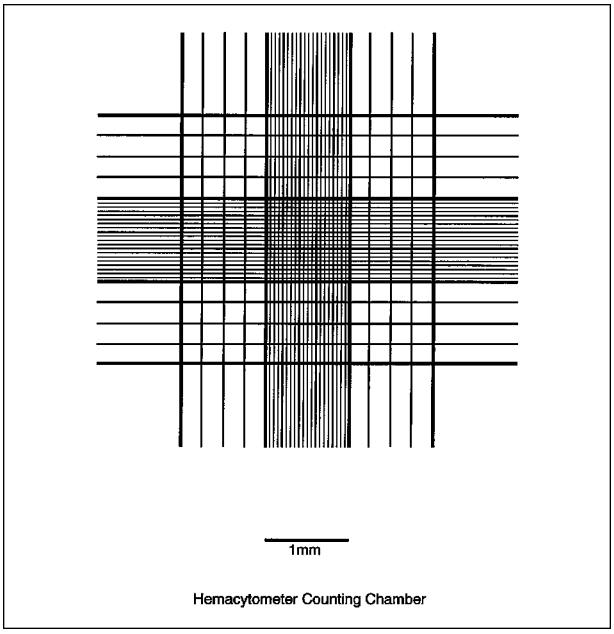

FIG. 8.7. *An Improved Neubauer Hemacytometer grid. Each hemacytometer has two grids. Sperm counts are determined by counting 5 of the 25 squares on one grid within the 1-mm square. A normal counting pattern is to count the contents of the following squares: upper left, upper right, lower left, lower right, and center. For the white blood cell count, the same pattern can be used, but count 5 of the 9 squares within the 3-mm square and total the number of cells on this grid and the second grid. Red blood cell counts are made by counting all cells within the 1-mm central square. To avoid overcounting cells that contact grid lines, include only cells that fall on the upper and left boundary lines of the square.*

TABLE 8.2B

White blood cell hematological values (average, standard deviation, and range) for healthy cranes of six species.[1]

	Whooping Crane N=17	Sandhill Crane N=5	Red Crowned Crane N=11	Sarus Crane N=12	Siberian Crane N=14	Wattled Crane N=6
White Blood Cell (N/µL)[2]	16999	13430	14933	14446	10770	12672
SD	6671	4626	4303	2687	2132	4761
Range	9752-29726	8392-20900	6327-23539	9072-19820	6506-15034	3150-22194
Heterophil (%)	56	53	41	59	53	48
SD	10	2	10	7	10	15
Range	38-74	50-56	22-62	45-73	33-73	21-81
Lymphocyte (%)	41	40	48	32	39	39
SD	11	3	9	8	10	20
Range	21-60	38-43	29-65	16-48	18-58	0-75
Monocyte (%)	2	6	6	4	3	5
SD	2	4	2	4	1	3
Range	0-6	3-13	2-10	0-12	2-6	0-11
Eosinophil (%)	1	2	5	5	5	8
SD	2	1	1	1	2	4
Range	0-5	0-3	3-7	3-7	1-9	1-17
Heterophil (N/µL)	9597	7078	6120	8486	5678	6108
SD	4616	2385	1859	1755	1030	1701
Range	4702-21997	4448-10868	2402-9838	4976-11996	3618-7738	2706-9510
Lymphocyte (N/µL)	6836	5353	7213	4571	4183	4890
SD	3064	2136	3051	1633	1814	4067
Range	3129-13506	3609-8987	1111-13315	1305-7837	555-7811	0-13024
Monocyte (N/µL)	390	802	882	606	370	604
SD	322	515	348	535	117	413
Range	0-976	334-1544	186-1578	0-1676	136-604	0-1430
Eosinohil (N/µL)	138	210	700	783	546	1070
SD	230	204	314	290	198	258
Range	0-688	0-418	72-1328	203-1363	150-942	554-1586

[1] *Basophils were absent in samples.*

[2] *As stated in the text, the White Blood Cell count is not derived from, and does not exactly equal, the totals for all WBC groups at the bottom of the table.*

12. The total WBC is determined as:
Total WBC/mm^3 = (total heterophils + total eosinophils)/(% heterophils + % eosinophils).

INTERPRETATION OF THE HEMOGRAM. Tables 8.2A and B have blood parameter reference ranges for five species of cranes. Similar values for other crane species are found in Connetta et al. (1974), Hawkey et al. (1983), Cook et al. (1989), and Puerta et al. (1990), and in the databases available through ISIS/MedARKS (see Chapter 10). Some additional parameters are not in the tables. Mean corpuscular volume is 1.12-2.58 × 10^{-3}mm^3 for Sandhill Cranes and 1.46-4.38 × 10^{-3}mm^3 for Whooping Cranes. Mean corpuscular hemoglobin is 3.2-9.8 μμg for Sandhill Cranes and 5.0-9.3 μμg for Whooping Cranes. The mean corpuscular hemoglobin concentration is 21.4-50.5% for Sandhill Cranes and 27.1-43.9% for Whooping Cranes. Table 8.3 provides hematologic changes seen in developing Sandhill Crane chicks. Reference values for these chicks are not the same as for adults.

A crane's blood cells generally respond to disease as do blood cells of other birds (Hawkey et al. 1983). For example, just as for other avian groups, anemia in cranes is associated with bleeding wounds, gastrointestinal foreign bodies, lead poisoning, blood

TABLE 8.3

Pediatric hematologic and serum chemistry values (mean; range) for captive Sandhill Crane chicks.[1]

AGE IN DAYS	0-2	6-8	13-15	20-25	27-29	34-36
Hematocrit (%)	33; 27-37	28; 23-30	28; 25-30	29; 25-33	28; 24-35	28; 25-32
Red Blood Cell Count (10^6/mm^3)	1.47; 1.22-1.71	1.31; 1.12-1.52	1.30; 1.09-1.48	1.32; 1.14-1.59	1.37; 1.18-1.65	1.32; 1.17-1.56
Mean corpuscular volume (MCV) (Fl)	217; 206-231	216; 187-235	206; 190-235	212; 195-235	208; 185-223	215; 192-235
White Blood Cell Count (10^3/mm^3)	20.5; 13.6-33.5	12.0; 7.0-48.3	16.0; 9.7-28.5	13.2; 7.8-19.4	12.9; 7.9-16.4	18.3; 8.3-29.1
Heterophil (%)	52; 38-77	52; 36-66	53; 36-75	53; 40-68	45; 24-61	43; 24-61
Lymphocyte (%)	40; 15-50	42; 30-59	45; 24-64	43; 28-49	50; 33-69	54; 35-76
Monocyte (%)	4; 1-8	5; 2-13	2; 0-6	4; 0-12	4; 1-11	3; 0-9
Eosinophil (%)	2; 0-4	2; 0-4	0; 0-1	1; 0-1	1; 0-2	0; 0-2
Basophil (%)	0; 0-1	1; 0-3	0; 0-1	1; 0-2	0; 0-1	0; 0
Total Protein (g/100mL)	3.4; 3.1-4.2	3.4; 2.9-4.5	3.5; 2.3-5.4	3.3; 3.0-3.6	3.4; 3.2-3.8	3.5; 3.0-4.0
Albumin (g/100mL)	<0.5	<0.5-0.5	<0.5	<0.5	<0.5-0.8	<0.5-0.5
Alkaline Phosphatase (IU/L)	179; 23-224	274; 186-372	331; 203-524	369; 230-538	391; 283-553	282; 327-510
Lactic Dehydrogenase (IU/L)	—	—	—	241; 204-288	271; 192-384	308; 293-322
Aspartate Aminotransferase (IU/L)	99; 88-110	262; 221-322	191; 143-263	144; 123-161	146; 118-160	151; 111-195
Glucose (mg/100mL)	—	—	228; 205-240	212; 178-251	241; 229-261	221; 204-237
Uric Acid (mg/100mL)	4.7; 3.7-6.9	5.5; 4.4-7.2	6.8; 6.4-8.3	7.6; 6.0-9.9	6.2; 4.8-9.3	5.4; 4.6-6.4
Gamma Glutamyl Transferase (IU/L)	—	—	—	2; 2-3	3; 2-3	3; 2-6
Creatinine (IU/L)	—	—	—	128; 62-252	144; 40-247	228; 140-290
Calcium (mg/100mL)	8.1; 6.0-10.6	6.9; 5.5-7.9	7.9; 6.8-8.8	8.0; 6.7-10.2	7.7; 5.7-9.9	8.9; 7.1-9.6
Phosphorus (mg/100mL)	—	—	5.8; 5.1-7.2	5.9; 4.5-8.1	6.1; 4.9-7.0	6.3; 5.7-7.0

destruction, or decreased blood cell production. An increase of basophilic appearing RBCs above 5% or a reticulocyte (a type of immature RBC) count above 10-15% is indicative of a regenerative anemia. Regenerative anemia is usually associated with blood loss or destruction from such causes as hemorrhage from wounds, blood parasites (causing RBC loss), gastrointestinal parasites, certain bacterial infections, lead, and other toxins. Non-regenerative anemia is associated with decreased RBC production from such causes as pesticide toxicity, lead toxicity, chloramphenicol toxicity, or certain viral infections.

High total WBC counts have been associated with chronic bacterial infections, avian tuberculosis, fungal infections, and stress. Low WBC counts are rarely seen, but have been documented in inclusion body disease of cranes, in overwhelming bacterial infections, and in immunologically depressed cranes.

Serum Chemistry

In addition to the information gained from examining the blood cells, there are many substances in the serum that can be analyzed to help assess the health of the crane patient. Serum chemistry reference

TABLE 8.3 CONTINUED

Pediatric hematologic and serum chemistry values (mean; range) for captive Sandhill Crane chicks.

Age in Days	41-43	48-50	55-57	62-64	69-76
Hematocrit (%)	29; 28-32	29; 28-31	28; 26-34	30; 28-34	34; 30-39
Red Blood Cell Count (10^6/mm^3)	1.30; 1.17-1.65	1.38; 1.19-1.62	1.34; 1.17-1.55	1.44; 1.29-1.73	1.57; 1.40-1.75
Mean corpuscular volume (MCV) (Fl)	211; 189-235	209; 191-223	210; 200-222	206; 196-223	214; 206-225
White Blood Cell Count (10^3/mm^3)	24.1; 14.5-32.8	23.6; 10.8-39.8	16.4; 10.8-23.9	14.7; 6.7-24.1	15.8; 5.6-20.4
Percent Heterophil	35; 20-61	42; 20-68	41; 21-64	40; 31-60	39; 18-49
Lymphocyte (%)	61; 38-77	56; 21-75	57; 31-73	60; 51-68	60; 49-83
Monocyte (%)	3; 2-5	4; 1-7	2; 1-6	3; 1-7	2; 1-3
Eosinophil (%)	0; 0-2	0; 0-1	0; 0	0; 0	0; 0
Basophil (%)	0; 0	0; 0	0; 0	0; 0	0; 0
Total Protein (g/100 mL)	3.0; 3.4-4.3	3.6; 3.3-3.8	3.7; 3.4-4.0	3.7; 3.1-4.1	3.8; 3.3-4.2
Albumin (g/100mL)	<0.5-0.6	<0.5-0.6	<0.5-0.5	<0.5-0.8	<0.5-0.8
Alkaline Phosphatase (IU/L)	449; 232-691	388; 286-529	399; 281-529	386; 271-616	288; 225-354
Lactic Dehydrogenase (IU/L)	239; 222-262	249; 194-292	285; 240-431	294; 192-499	324; 176-472
Aspartate Amino-transferase (IU/L)	159; 151-161	159; 110-184	152; 133-177	152; 119-182	173; 152-199
Glucose (mg/100mL)	228; 219-259	219; 214-233	214; 196-228	223; 197-254	212; 202-222
Uric Acid (mg/100mL)	6.1; 5.9-6.4	5.4; 4.7-8.1	5.2; 5.0-5.3	5.6; 4.8-8.4	6.4; 4.7-8.0
Gamma Glutamyl Transferase (IU/L)	3; 3	3; 2-5	2; 2-4	2; 2-4	2; 2-3
Creatinine (IU/L)	159; 128-191	168; 152-184	127; 92-169	172; 81-306	169; 71-337
Calcium (mg/100mL)	8.9; 6.1-9.3	8.0; 6.9-9.2	9.3; 8.2-10.2	8.9; 6.4-11.0	9.2; 7.1-10.9
Phosphorus (mg/100mL)	6.9; 5.9-7.9	6.5; 5.4-7.1	6.8; 6.0-7.7	6.8; 6.0-7.8	6.9; 6.0-7.8

[1] *Based on unpublished work from ICF*

ranges for cranes are given in Tables 8.2 and 8.3, and in the same sources cited earlier for blood parameters plus Chappell and Brannian (1984). The serum chemistry tests described below can be performed on the automated analyzer used for mammalian and human serum chemistry analysis in a medical laboratory. Alternately, many veterinary facilities have analyzers. The actual test procedures will vary with the analyzer used and should be handled by the technical staff familiar with the operations of the machine.

Useful crane serum parameters include total protein, albumin, calcium, glucose, lactic dehydrogenase, alkaline phosphatase, aspartate aminotransferase (glutamic-oxaloacetic transaminase), uric acid, creatinine kinase, creatinine, bile acids, sodium, potassium, calcium, and phosphorus. Other serum chemistries, important in human medicine, such as blood urea nitrogen and alanine aminotransferase (glutamic-pyruvic transaminase) are not as useful for diagnosis of crane diseases. The alterations in serum chemistry seen in sick cranes are not well documented in the literature, but are believed to be similar to what has been recorded for other avian species (Hochleithner 1994).

Total protein (normalsfor all chemistriesare listed in Table 8.2A) values canbe elevatedwith dehydration, lipemia,or hyperglobulinemia secondary to chronic diseases (such as aspergillosis, avian tuberculosis, and chronic bacterial infections such as *Staphylococcus*). Total protein valuescan decreasein malnutrition, acute infections, chronic liverdisease, ormalabsorption ofnutrients caused by severe intestinal parasitism.

Calcium values increase with egg laying and can be as high as 3 times normal. Decreased calcium values are seen with egg binding or nutritional imbalances. Low calcium levels have been documented as leading to seizures in some avian species, however, this has not been reported in cranes. The calcium/phosphorus ratio is important, especially in growing birds, and should be 2:1.

Glucose increases with stress or diabetes mellitus (a disease reported in other avian species but not in cranes to date). Decreases in serum glucose are seen in starvation, septicemia, severe liver disease, endocrine disorders, or with improperly processed samples. If the RBCs remain with the serum over 60 min at 22° C (70° F) or higher, the cells continue to use glucose from the serum, lowering the glucose reading.

Lactic dehydrogenase (LDH) levels in birds can be increased by liver disease, aminoglycoside therapy, intramuscular injections, cardiac or skeletal muscle catabolism, or *Chlamydia* infections. Decreases do not suggest any specific condition. In addition, increased LDH values can result as an artifact if the serum sample is hemolyzed. High levels of this enzyme are present in the liver, skeletal muscle, and heart, while moderate amounts are present in kidney tissue and small amounts in intestine based on studies in other avian species. Any process or disease affecting these tissues may result in serum elevations of the enzyme.

Aspartate aminotransferase (AST) can be increased in *Chlamydia* infections, liver disease, bacterial septicemia, soft tissue trauma, starvation, toxicity, neoplasia, aminoglycoside therapy, or intramuscular injections. Decreases have no clinical significance. The enzyme is found in the liver, heart, brain, lung, bone, and muscle. Any process affecting one or more of these tissues may cause the enzyme to be elevated. In cranes, elevations are common due to mild muscle damage associated with normal handling. If the creatinine kinase (CK) is also mildly to moderately elevated, then mild, reversible, muscle damage is probably the cause. Severe elevations of both (i.e., >2 times normal value) indicate a serious muscle disease such as exertional (capture) myopathy. If the AST is elevated and the CK is not, then liver disease is a possibility.

Creatinine kinase will increase in muscle trauma (including intramuscular injections), central nervous system disorders, cardiac disease, or lead poisoning. Decreases are not significant. Because CK increases in heart and muscle disease, but not in liver disease, it is important to test with AST and LDH. Elevations in AST and LDH, but not CK, suggest liver disease or *Chlamydia* infections, whereas elevations in all three generally indicate heart or muscle disease.

Uric acid is the primary excretion product of the kidneys from nitrogen (protein) break down in cranes. Increases are characteristic of renal disease (including gout and damage from aminoglycoside medications), but may also occur in dehydration, starvation, trauma, or neoplasia. Decreases are not considered significant.

Creatinine may increase with renal disease, septicemia, egg peritonitis, or high protein diets (the latter occurs in some avian species, but has not been documented in cranes). Decreases are not considered

significant. The test is not very sensitive and some authors consider it of limited diagnostic value, but elevations have been seen in cranes with severe renal disease.

Bile acids are only elevated by liver disease and are considered the best serum indicator of a liver disorder. Lower than normal levels of bile acids are not considered significant. Bile acid levels tested in cranes at ICF have generally been lower than 80 µmol/L.

Alterations in the electrolytes sodium (Na), potassium (K), and chloride (Cl) indicate a serious change in the bird's acid/base balance and metabolic state. In a crane, this is most likely due to renal disease (increased K and decreased Na), diarrhea (decreased K and Cl), or shock acidosis (increased K and decreased Na). An elevated K level by itself is often due to hemolysis of the serum sample.

Parasitology

Intestinal parasites can be diagnosed at necropsy and by sampling fresh fecal samples in living cranes. Three types of fecal examination (direct smear, flotation, and sedimentation) are routinely performed.

Flotation is a practical technique because it helps to concentrate the eggs of the parasites and removes other material in the sample. The eggs of nematodes, cestodes, and acanthocephalans float as do oocysts of coccidia, the cysts of *Giardia*, and other protozoa. Examine an adequate amount (ca 1-2 g) of fresh feces. Nematode eggs will larvate and *Giardia* may perish in samples that have lain on the ground more than 15 min. Feces lying on the ground can also be invaded by free-living, non-parasitic, nematodes which can confuse results.

The diagnostic techniques are most valuable if standardized procedures (described in Greiner and Ritchie 1994 and various parasitology texts) are used. The standard flotation medium is saturated sodium nitrate (568 g/L water). However, Sheather's sugar solution (500 g table sugar, 320 mL water, 6.5 g phenol crystals) works best to detect coccidia oocysts. Saturated zinc sulfate (336 g/L water) works well for *Giardia* concentration and detection (Greiner and Ritchie 1994). The fecal specimen is mixed with at least 10 times the volume of flotation medium. The mixture can then be strained and placed in a small cylindrical container. The flotation medium should fill the container. A microscope coverslip is placed on top of the container and should make contact with the flotation medium. The mixture should be allowed to stand 45 min before the coverslip is placed on a microscope slide and read. To speed up the process, the strained fecal/flotation medium mixture can be placed in a 15 mL centrifuge tube and spun at 1200 rpm for 10 min. Then the top layer of medium is collected and the drop placed on a microscope slide, covered with a coverslip, and read. Parasites commonly diagnosed from eggs seen on flotation include *Capillaria* sp., *Eucoleus* sp., ascarids, acanthocephalans (*Macracanthorhynchus* sp.), and gapeworms (*Syngamus* sp., *Cyathostoma* sp.). Oocysts of *Eimeria* are also seen.

Direct smears are useful for detecting *Giardia*, coccidia, and other protozoan parasites such as *Hexamita*. Samples must be fresh. A small amount of feces is mixed (1:2) with normal saline or lactated Ringer's solution on a microscope slide. A coverslip is placed on top, and the slide is read under the microscope.

Sedimentation is the only technique that will detect fluke eggs, but it can also detect nematode eggs. A sample of feces is mixed with 1% liquid soap in water. Remove the supernatant after 5 min, refill the tube with soap and water, mix, and let stand another 5 min. Again, remove the supernatant, then spread the sediment on a microscope slide, place a coverslip on top, and read.

Generally, a low power objective (10x) is used for scanning the microscope slide. The higher power objective (40x) can be used to examine individual eggs or oocysts. The entire area of the sample on the microscope slide should be scanned for each sample, because some parasites only produce small numbers of eggs, and the presence of even one egg is diagnostic. There is no direct relationship between egg counts and number of adult parasites present. However, if samples are taken before and after treatment, egg counts are useful in providing information on the effectiveness of antiparasitic medications. The significance of intestinal parasitism for cranes is discussed under Parasitic Diseases later in this chapter.

Although hematozoa (blood parasites) are not common in captive cranes in North America, all blood smears should be scanned for these protozoa. Thick smears are better than thin smears for this purpose. Routine hematology stains are sufficient for screening, but more specific techniques (Olsen and Gaunt 1985) are recommended.

Radiology

The radiographic examination is an additional diagnostic aid. All veterinarians working with cranes should have access to radiology equipment. Radiography techniques used for other avian species or for extremities of mammals can be adapted for cranes. Techniques are available in veterinary and radiology texts and are reviewed in McMillan 1994. Radiographic manifestations, even under normal conditions, vary with species of crane, age, sex, time in captivity, and even method of flight restraint (i.e., non-flighted birds normally have atrophied pectoral and wing muscles). Traumatic injuries to the long bones and skull are the most frequent injuries seen in crane radiographs. Radiographs are also important for diagnosis of soft tissue diseases and ingested foreign bodies.

Radiology Protocol

Each institution will need to develop its own exposure chart based on its particular machine, screen, and film. Start with a general bird or cat technique chart, and use the appropriate body measurements taken from the crane being radiographed. From a log book that records patient identification, species, date, body part, positioning, measurement, exposure technique used, and comments on the technical quality of the film, you will be able to develop an exposure technique chart for cranes.

It is often preferable to use general anesthesia (optimally isoflurane gas) for radiography of cranes. Having the crane anesthetized will decrease the stress for the bird, decrease movement, and improve positioning resulting in a better quality radiograph. If anesthesia is not available or indicated, hooding the crane may be helpful. Physical restraint (without anesthesia) is routinely used for radiographs of some body areas such as legs or feet, or for quickly scanning the gastrointestinal tract (e.g., looking for ingested metal).

For an unanesthetized crane, use the shortest exposure time to minimize any problems with motion. Therefore, a high kilovolt potential (kVP) and the highest milliampere (mA) setting is used to compensate for a short exposure time. When radiographing an anesthetized crane, there is more leeway in adjusting settings to obtain the best quality radiograph for diagnostic purposes. Higher kVP settings provide for a longer scale of contrast and more exposure latitude. With lower kVP settings, more subtle changes in subject density can be seen on the radiograph.

Whenever possible, take two or more views (Fig. 8.8). Normally these are dorso-ventral and lateral radiographs, but oblique images are sometimes helpful in enhancing the diagnostic value of the radiographs. However, with some body parts such as the wing of a crane, positioning can be very difficult, and the information gained from the second view may not always be worth the additional time and handling.

All but essential personnel should leave the room. Anyone present in the room should wear a lead apron, neck shield and gloves, and have an exposure monitoring badge.

To obtain a good diagnostic radiograph, **develop** it properly. If an automatic film processor is used, feed the film in the darkroom and wait for the finished film. To manually process a negative:

1. Extinguish all lights except a safe light (red) when handling unprocessed film.
2. Touch the film only by the corners as it is taken from the cassette and attached to the appropriate size hanger.

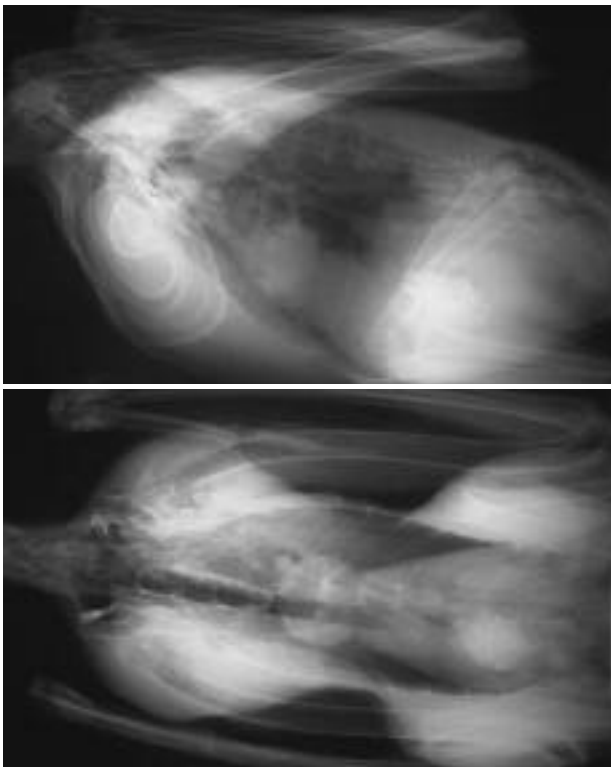

FIG. 8.8. *Lateral and dorsal-ventral view (radiograph) of normal Whooping Crane.* PHOTO GLENN H. OLSEN

3. Refill the radiograph cassette with the appropriate size of unexposed film. Make sure the unused film is returned to the stock box and resealed against light.
4. Take the hanger with the exposed film and place it in the developing solution. Turn on the pre-set timer. Periodically agitate the hanger gently during developing.
5. When the time has expired for developing, remove the hanger and film, and dip it several times in the wash solution (usually water). Then allow most of the wash to drain.
6. Place the hanger in the fixer solution for twice the length of time used for developing. Occasionally agitate.
7. If it is necessary to view film before drying, remove film from the fixer after the first minute and examine.
8. Remove the hanger and film from the fixer when time has expired, and place it in the wash (water) for twice as long as the time in the fixer. Then allow the film to dry on the hanger.

When storing radiographs, remove the corners with scissors so that the small puncture holes created by the hanger do not scratch other film in the envelope.

Radiographic Interpretation

Accurate interpretation of radiographs depends upon: (1) the case history of the crane, (2) findings from the physical examination, (3) radiograph quality, (4) density of the tissue or object being studied, and (5) proper positioning of the patient (take at least two views). Special techniques, such as the use of contrast media, can also help increase the diagnostic value of radiographs. Knowledge of normal anatomy is important in diagnosing abnormalities; it is helpful to keep a reference collection of radiographs of normal cranes of different species and ages for comparison. ICF has produced radiographic series on the developing legs of three species of crane chicks (Siberian, Whooping, and Florida Sandhill Cranes). Copies of these films are available at cost from ICF.

Radiographscanevenbeusedfornon-dense **carcinomas**. Osteosarcomasoccurrarelyincranes. Typical radiographicevidenceincludesosteoscleroticareas, osteolysis,orperiostealboneformationwithsomebone formingwithinthesofttissue.Chondroma-likelesions have beenseeninwild Florida Sandhill Cranesandone Whooping Crane(M.G. Spalding, Universityof Florida, Gainesville, Florida,personalcommunication).

Infectious Diseases

Bacterial Diseases

Diseasesofbacterialoriginare commonlyencountered incranes. Stresscancontributetotheoutbreakand spreadofbacterialdiseases(Carpenter 1986). *Salmonella* spp.,including a varietyofserotypes,onebeing *S. typhimurium*, have beenisolatedfromthefecesof cranes (Windingstadetal. 1977; Langenbergand Dein 1992).Theoriginandsignificanceoffecal *Salmonella* isolatedfromclinicallyhealthybirds isnotknown. However, *Salmonella* cankillchicks,mayaffectfertility, andis a concerninbirdsfor releaseintothewild. For these reasons,aneffort isoftenmadetostoporcontrol shedding by *Salmonella* carriers. Frequently, theseinfectionsare transientandself-limitingwithtemporary isolationandsurveillanceallthatisnecessary. However, someinfectionsare persistentandantibiotictherapyis useful. Birdstreatedwithampicillin,tetracycline, trimethoprim-sulfa,ormetoprim-sulfaantibiotics, orwithanexperimental *S. typhimurium* bacteria,(see Table 8.1 fordosages)have testednegative onseveral subsequentcloacalcultures.Antibiotictherapyis controversialbecauseitmay onlytemporarilystop sheddingorevencause a permanentcarrierstate.

Escherichiacoli is routinelyculturedfromthegastrointestinaltractofboth youngandadultcranesinlow tomoderatelevelsandcanbeconsiderednormal flora. Evencranechicksraisedindoorsoncarpetingwillhave *E. coli* aspart oftheirgastrointestinal flora by day 6 posthatch. However, *E. coli* canbepathogenicandhasbeen thecauseofdiarrheaandsubsequentdeathin young chicks(seeChapter 5, Veterinary Techniquessection).

Mycobacterium avium infections (avian tuberculosis) are widely reported in captive cranes. In fact, some feel that cranes are more susceptible than many other avian groups to this usually fatal infection found in the gastrointestinal tract, liver, spleen, and other internal organs. Clinical signs include weight loss, anorexia, abdominal organ enlargement, the presence of masses on radiographs, and an elevated WBC count. Diagnosis can be difficult: the tuberculin skin test used in poultry does not work in cranes, so a combination of laparoscopy, liver biopsy, and fecal culture are often used (Langenberg and Dein 1994). Treatment using ethambutol and rifampin have been attempted in two cases (Snyder and Richard 1994), but the success rate is not yet clear.

Other bacteria isolated from cranes include *Pasteurella multocida*, *Arizona* spp., *Clostridium* spp., *Erysipelothrix* spp., and various Enterobacteriaceae such as *Pseudomonas* sp. and *Klebsiella pneumoniae*. See Table 8.1 for suggested antibiotics and dosages for treatment.

Respiratory Diseases

The bacterial agents most often associated with pneumonia in cranes include *Pasteurella multocida*, *Yersinia pseudotuberculosis*, *Escherichia coli*, *Klebsiella pneumoniae*, *Pseudomonas aeruginosa*, *Salmonella typhimurium*, and *Streptococcus* spp. Cultures can be obtained from the trachea or choana, from sinus exudate or air sac wash, or by air sampling in fledgling cranes (Shane et al. 1986). Even a positive bacterial culture may not diagnose a cause; it is possible that the bacteria are secondary to a viral infection or associated with another respiratory problem such as aspiration or scoliosis. Viral diseases including Newcastle disease (paramyxovirus group 1) are possible. Fungal infections, especially with *Aspergillus fumigatus*, are commonly seen in cranes although usually as a secondary problem after treatment for bacterial pneumonia or in a generally debilitated bird. Fungal infections are difficult to treat, and are often diagnosed when a crane fails to respond to standard antibacterial therapy. Although buccal contamination of cultures is normal, the first step in the treatment of a respiratory disorder is culturing for bacteria and fungi. Radiographs can also be helpful in diagnosis.

Maintain the sick bird in a non-stressful, warm (70^0-85^0 F, 21^0-29^0 C) environment. Hospitalization is recommended for the first 7-10 days. Supportive care initially consists of systemic antibiotics. Gentamicin, trimethoprim/sulfadiazine, amikacin, piperacillin sodium, or enrofloxacin (see Table 8.1 for dosages) are all recommended as initial choices until the results of sensitivity tests from bacterial cultures are received. Other possible treatments include intramuscular (IM) injections of vitamins A, D, and E; intravenous (IV) or subcutaneous (SQ) fluids; and supplemental feedings if weight falls. If aspergillosis is suspected or confirmed by culture, use antifungal therapy such as fluconazole (oral), itraconazole (oral), amphotericin-B (intravenous or intratracheal), or a combination of the latter with one of the former. Tracheal flushes and nebulization are also recommended (see Table 8.1 and 8.4). Our best results in nebulizing chicks have come from clotrimazole, first used in 1995.

TABLE 8.4

Medications used for nebulization of cranes.

MEDICINE	INDICATIONS	DOSAGE
Enrofloxacin	Antibacterial	22 mg in 10 mL saline
Gentamicin	Antibacterial	50 mg in 10-30 mL saline or distilled water
Amikacin	Antibacterial	50 mg in 10 mL saline
Tylosin	Antibacterial	100 mg in 10 mL saline
Erythromycin	Antibacterial	200 mg in 10 mL saline
Polymyxin B	Antibacterial	333,000 U in 5 mL saline
Sulfadimethoxine	Antibacterial	200 mg in 15 mL saline
Piperacillin sodium	Antibacterial	200 mg in 10 mL saline
Amphotericin B	Antifungal	5-10 mg in 15 mL water[1,2]
Clotrimazole	Antifungal	30 mg, do not dilute[2]
Acetylcysteine[3]	Mucolytic	0.25-1.0 mL 10-20% solution in 10-15 mL saline or distilled water

[1] *Can use distilled or sterile water for injection.*

[2] *Use disposable pediatric nebulizer and O_2.*

[3] *Can be mixed with other medicines.*

Nebulizationtherapy withantibioticsisbestaccomplishedwithanultrasonic nebulizer commonlyusedin human respiratory therapy(see Appendix).Thisunitis usedwith a mobilebacterial filterto remove bacteria fromtheairsupply. Ideally, theunitshouldhave an adjustableoutputchamberfrom 0-7.5 mLandhave an alert signaltoindicatewhenthechamberisempty. The fanshouldhave variablespeedcontrolandaerosol output.Thenebulizationtherapyshouldtotal 1 hour daily, dividedinto 2-3 equaltreatments. A 1:10 or 1:20 mixture ofgentamicin (orother watersolubleantibiotic)tosalineortodistilledwaterisused(see Table 8.4).Thecranemustbehousedin a relativelyair-tight chamber, suchas a Snyder oxygencage,duringtherapy. Nebulizationtherapywithantifungalagents(see Table 8.4) usually requires a pediatricnebulizerandforcedair supplied by a tank(O_2) oraircompressor.

Nebulization therapy is effective because it brings the antibiotic/antifungal directly to the affected respiratory membrane. Because there is usually little or no uptake of drugs into the crane's circulatory system from this application, standard systemic antibiotic therapy must still be used. The humidification of the respiratory epithelium during nebulization is soothing for the patient. In addition, a mucolytic agent such as acetylcysteine (0.25-1.00 cc) can be added to each 10-15 cc of nebulization fluid to help break up mucous in the respiratory tract.

Viral Diseases

Several viral diseases have been identified in cranes including avian pox (Simpson et al. 1975) and Newcastle disease (Kaleta and Marschall 1981). Inclusion body disease of cranes (IBDC, crane herpes virus) and eastern equine encephalitis (EEE) have had a major impact on captive cranes (Carpenter et al. 1987, 1989; Dein et al. 1986). The reovirus seen in Grey Parrots (*Psittacus erithacus*) (Graham 1987) is similar to a reovirus that caused chick mortality at Patuxent.

In 1978, IBDCcausedthedeathof 18 cranesof 4 speciesatICF(Dochertyand Henning 1980). Signs were nonspecific butincludedanorexia,lethargy, weakness,anddyspnea.Thepathologywastypical ofherpes virusinfectionswithaninclusion bodyhepatitisand splenitisprogressingrapidlytodeath.Liverandspleen are thebesttissuesforvirus isolation.Thestatusof IBDCinwildcranesisuncertain.Limitedserotesting onsixcranespeciesfrom NorthAmerica, Eurasia,and Africahasfoundonlytwopositive Eurasian Cranesin southwest Russia. However, similar *Herpes* viruseshave beenisolatedfromillcranesin Austria, France, Japan, China,and Russia (Carpenter 1993 andICFunpublisheddata).The IBDCvirusmaypersistin a dormant stateinaninfectedbird makingdetection by virus isolation difficult. Serologicaltesting forIBDCantibody(avirus-neutralizationtest)isavailableonlyat the National Wildlife HealthCenter, Madison, Wisconsin. There isnospecific treatmentforthisdisease. Because ofthedangerposed by thisvirustoothercranesin a collectionorinthewildandthepotentialfor a carrier state,itis usuallyadvisabletoeuthanize orcompletely isolateinfected cranes. Trans-ovariantransmissionof thevirushasnotbeenseentodate,soeggsmaybe safelytakenfrom antibody-positive cranes.

The EEE virus, an arbovirus (arthropod borne virus), is native to the eastern and north-central North America, parts of Central and South America, and the Caribbean Islands. The virus is primarily carried by the mosquito *Culiseta melanura*, a species which breeds primarily in hardwood swamps. Nearly all exposed native birds develop antibody titers with no morbidity or mortality. However, some Whooping Cranes and some species introduced into North America often develop clinical signs and many die.

In 1984, EEE virus killed 7 Whooping Cranes at Patuxent (Dein et al. 1986; Carpenter et al. 1987, 1989). Three of the 7 birds showed lethargy, ataxia, and neck and leg paresis, while 4 showed no clinical signs. Of the 32 surviving Whooping Cranes in this captive flock, 14 (44%) developed antibodies to EEE. Subsequent to the 1984 outbreak, an inactivated EEE vaccine for humans was found to stimulate antibody titers in both Sandhill and Whooping Cranes (Clark et al. 1987). Since 1985, all Whooping Cranes held in areas where the disease is present have been vaccinated. In addition, there is a screening program at Patuxent to detect EEE by examining mosquitos and by monitoring exposure in sentinel Bobwhites (*Colinus virginianus*) and Sandhill Cranes (Pagac et al. 1992). This program documented an EEE incident in 1989 where the virus was present in mosquitos and spread to the quail, but no losses were recorded among 55 exposed but vaccinated Whooping Cranes, leading us to conclude that protective titers were established by the vaccination program. A variety of crane species at zoos have been vaccinated with commercial equine EEE vaccines with no reported ill effects. However, Clark et al. (1987) reported that two species of cranes had a poor antibody response to equine EEE vaccines.

Parasitic Diseases

Parasites opportunistically infect stressed or crowded cranes during migration or when in captivity. Clinical signs of parasitism are usually non-specific and may include weight loss, lethargy, diarrhea, or dyspnea. Heavy parasitic infections can also cause malnutrition and increase susceptibility to other diseases (Carpenter 1979).

A parasite monitoring and control program is critical to maintain and breed healthy cranes in captivity. Therapeuticand prophylactic administration of parasiticides (Table 8.5) is an important part of any medical program. In addition, reducing pen crowding, practicing pen rotation, cleaning facilities, quarantine and treatment of new birds, and separating birds by age are important parts of a control program. Prophylactic administration of anti-parasite medications is especially useful with young chicks (see Chapter 5), cranes being prepared for release or shipment to other collections, and when introducing cranes into new (parasite-free) facilities.

Cranes can be infected by a number of species of protozoa including the blood-born parasites *Hemoproteus* and *Leucocytozoon*, but the true significance of these infections has not been documented. *Hexamita* has been implicated as the cause of enteritis and death of captive Florida Sandhill Cranes (M. G. Spalding, University of Florida, Gainesville, Florida, personal communication).

Themostcommonandbest-documentedprotozoanparasitesofcranesare thecoccidia.Both *Eimeria gruis* and *Eimeria reichenowi* (Fig. 8.9a)are common parasitesofWhooping, Demoiselle,and Sandhill Cranes,andare thelikely speciesfoundinother cranes. In addition, *Adelina* sp. hasbeenfoundin Sandhill Cranes(Courtneyetal. 1975). *Isospora lacazei* hasbeen foundintwocaptive Whooping Cranes, butwas thoughttohave beenduetocontamination offood by fecalmatterfrompasserines (Forresteretal. 1978).

Cranes,likeseveralotherbirdsgroupsincluding geeseandturkeys,have anextra-intestinal formof parasitism by *Eimeria* inadditiontothegastrointestinalinfection(Carpenter 1993). In thisextra-intestinal form,calleddisseminatedvisceralcoccidiosis(DVC), endogenousstagesoftheparasitedisseminatefromthe alimentary tractthroughoutthebodycarried by the bloodorpossibly lymphaticsystems.This resultsin generalinflammationoforgans, seenasbroncho-pneumonia,hepatitis, myocarditis,splenitis,andenteritis, inadditionto theformationofdiscretegranulomatous nodulesinanyoftheseorgans.Thediseasecanbe devastatingincranechicksunder 60 daysofage. It is a seriousproblemat Patuxent,buthasnotbeendocumentedatICFwhere winters are muchcolder.

In captivity, concentrations of *Eimeria* inthe soilare oftenmuchhigherthanwouldbefoundinthe wild. Theparasiteshave been documentedinwildcranes, butthe roleof DVC insurvival ofwildcraneshasnot been studied. In captive colonies,several anti-coccidial medications (includingamproliumandmonensin, see Table 8.5) have proveneffective incontrollingthe incidenceof DVC whentheyare continuouslymixed withthe food(Carpenteret al. 1992). Intestinal coccidiainfectionsinindividualbirdshave beensuccessfullytreatedwith amprolium,trimethoprim-sulfa, ormetoprim-sulfa,andsulfadimethoxine.

Endoparasites, including acanthocephalans, cestodes, trematodes, and nematodes, have been found in cranes (Carpenter 1993). The overall effect of such parasites on both wild and captive cranes is not well documented. Acanthocephalans (spiny headed worms; Fig. 8.9b) occasionally cause perforation of the intestines leading to peritonitis and subsequent death in captive crane chicks. This parasite's significance in older captive birds and in wild birds is unknown. No known treatment is available for this parasite. Because the earthworm (*Lumbricus* sp.) may be an intermediate host, rearing chicks indoors may prevent infection.

Gapeworms (*Syngamus* sp. and *Cyathostoma* sp.) have caused severe tracheitis, bronchitis, and obstruction of the trachea through irritation and formation of mucous plugs, leading to death. Signs include dyspnea and open-mouth breathing (gaping). Sometimes gapeworms can be seen in the upper trachea of a symptomatic bird. Additionally, diagnosis is possible by examining tracheal washes and occasionally by fecal flotation. Ivermectin or fenbendazole (see Table 8.5 for dosages) have been effective in eliminating the parasite. Pen rotation will help prevent reinfection. Earthworms carry gapeworm eggs, therefore decreasing a crane's exposure to earthworms will help control infection rates.

Capillarids (*Capillaria* sp. and *Eucoleus* sp.; Fig. 8.9c) and *Ascaridia* sp. (Fig. 8.9d) can cause debilitation and occasionally contribute to death. Both are readily diagnosed on fecal flotation. Treatment with ivermectin or fenbendazole (see Table 8.5 for dosages) is effective, but reinfection is frequently possible unless birds are moved at least annually to a fallow pen. Treatment success has been highest when both ivermectin and fenbendazole are used concurrently.

TABLE 8.5

Antiparasitic medications used in cranes.[1]

Drug	Indications	Route of Administration	Dosage	Treatment Schedule Per Day
Anticoccidials				
Amprolium	Anticoccidial	Food	0.0125 mg/kg (Prophylactic) 0.025 mg/kg (Therapeutic)	Continuous for 2 weeks, minimum
Amprolium	Anticoccidial when other forms of this drug are not appropriate	Drinking water	0.006%	Continuous
Monensin sodium	Anticoccidial	Mixed in feed	90 ppm	Continuous or seasonally
Triple Sulfa Soluable Powder[2]	Used when clinical evidence of coccidiosis	Drinking water	1.5 tsp/gal	2 days on; 3 days off; 2 days on; 2 days off; 1 day on
Trimethoprim sulfa	Used when clinical evidence of coccidiosis	Oral IM	See Table 8.1	1-2/day
Ormethoprim sulfadimethoxine	Used when clinical evidence of coccidiosis	Food	0.015% ormethoprim 0.026% sulfa	continuous for 3 weeks
Sulfa dimethoxine	Used when clinical evidence of coccidiosis	Oral	50 mg/kg	1/day for 2 weeks
Antinematodals				
Fenbendazole	Capillariasis, other nematodes	Oral	100 mg/kg	5 days, then repeat in 10-14 days
Ivermectin	Broad-spectrum	IM	0.2 mg/kg	2 doses 10-14 days apart or as needed
Levamisole	Safe, efficacious broad-spectrum anthelminthic	Oral	40 mg/kg (25 mg/kg for chicks)	Bi-weekly or as needed
Piperazine	Treating individuals or groups of cranes for ascarids	Drinking water	15-20 g/gal	3 days; repeat in 2 weeks
Pyrantel pamoate	Intestinal nematodes	Oral	4.5 mg/kg	2 doses 10-14 days apart
Thiabendazole	Wide range of antiparasitic action with a high degree of efficacy and safety	Oral	100 mg/kg	Weekly or bi-weekly as needed
Anticestodals and Antitrematodals				
Albendazole	Effective in treating some trematodes	Oral	20 mg/kg	2 doses 1 week apart
Praziquantel	Effective in treating cestodes; potentially toxic	Oral	6 mg/kg	Bi-weekly or as needed
Ectoparasiticals				
Carbaryl	Control of most ectoparasites	Topical	5% powder	Weekly or bi-weekly or as needed
Pyrethrins	Control of most ectoparasites	Topical	0.10% powder	Weekly or bi-weekly or as needed

[1] *Based on Olsen and Carpenter 1996.*

[2] *Active drug ingredients: sulfamerazine sodium 27.20%, sulfamethazine sodium 27.20%, and sodium sulfathiazole sesquihydrate 29.85%.*

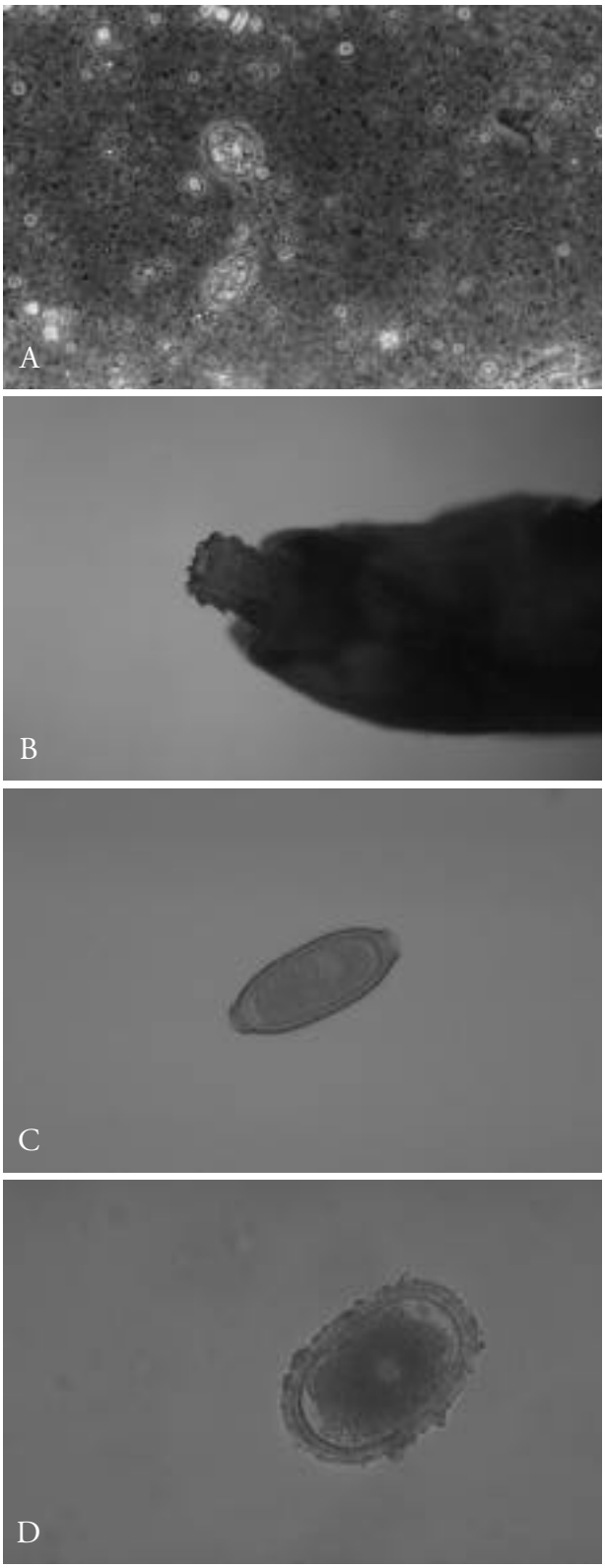

FIG. 8.9. *Photomicrographs of parasites: A, ova of Eimeria gruis (pyriform, 10 × 14 μ) and Eimeria reichenowi (round, 11 × 13 μ); B, head of acanthocephalan (proboscis 0.5 mm); C, Capillaria egg (25 × 50 μ); D, Ascaridia egg (50 × 90μ).*
PHOTOS HARRY DANFORTH (A) AND GLENN H. OLSEN (B-D)

Ectoparasites including 5 mite species (Order Acarina) and 4 biting lice species (Order Mallophaga) (Forrester et al. 1976; Atyeo and Windingstad 1979) are seen in cranes. No pathological significance, except possibly in young or debilitated birds, has been noted in ectoparasite problems. A dusting with 5% carbaryl or 0.10% pyrethrins (Table 8.5) is very effective as a treatment. In addition, dusting can be done during an annual health check as a prophylactic measure.

Problems with biting and stinging insects such as black flies (*Simulium* sp.), bees (*Apis* sp.), and wasps (*Vespis* and others) sometimes cause minor skin irritation, excessive preening, and behavioral signs of discomfort and stress. At ICF, equine insect repellents containing pyrethrins or carbaryl have been somewhat effective (see also Chapter 11F).

Non-Infectious Diseases

Trauma

The most frequent causes of trauma will vary between institutions, but include collisions with pen structures and during capture, handling, and shipping. Even with good husbandry and excellent facilities, aggression associated with establishing dominance hierarchies, mate selection, and defense of territory, food, or water remains important (see Chapter 6; Carpenter et al. 1976; Carpenter and Derrickson 1981). **Dangerous situations** are: (1) pair formation, especially in a community (group) pen with 3 or more birds, (2) when introducing new cranes into an established social group, and (3) the escape of a crane into a neighbor's pen. Generally, the intruder will be the victim of the aggression. At Patuxent, 7.3% of the Whooping Crane deaths occurring during the 15 year period from 1966 to 1981 were from aggression (Carpenter and Derrickson 1981).

Most aggression-related injuries are to the neck and head (Fig. 8.10). Depending on how soon the crane is found, the victim will often be in a deep state of shock. **Standard shock treatment** is given: corticosteroids, fluids (IV and SQ), and antibiotics (Table 8.1) are administered. The wounds are cleaned, and following stabilization, the bird is radiographed to assess skull damage. Supportive care, especially during the first 24 hours, is critical. Patients that survive the initial trauma and the first 48 hours normally make a complete recovery, but many cranes carry scars for life.

Fig. 8.10. *Head and neck injuries due to aggression.*
Photo David H. Ellis

Occasionally, a parent will attack a chick (see Chapter 5), but not normally without an underlying cause, such as chick lethargy, the parent reacting to some disturbance, or the parent not being prepared for a chick. Normally such chicks are killed.

An important source of trauma for captive cranes is collisions with fences and buildings. Anatomically and behaviorally, cranes seem predisposed to damage their long legs, beaks, necks, and wings. Intensive management of cranes for production can exacerbate this problem. Careful attention to pen and facilities design (see Chapter 12) and capture, handling, and shipping techniques (see Chapter 2) will significantly reduce trauma cases.

Trauma to the neck, head, or beak is second only to leg and wing injuries. Neck injuries generally occur when a crane runs or flies into a pen fence or building wall. The resulting damage to the cervical vertebrae and spinal cord is often fatal, but if not, it often results in signs of ataxia, paresis, or abnormal neck position. Treat for shock using corticosteroids (dexamethasone has been used successfully in several cases; see Table 8.1 for dosage). Methocarbamol (see Table 8.1 for dosage), a muscle relaxant, was useful in one successful case (Done et al. 1993).

Ocular injuries that have been seen in captive cranes include lacerations of the third eyelid and the primary lids, and corneal lacerations, abrasions, and punctures. These injuries can be repaired with standard ophthalmic surgical procedures described for birds and mammals (Magrane 1965; Karpinski and Clubb 1986; Murphy 1987). Prophylactic treatment with aminoglycoside ophthalmic antibiotics is recommended to prevent secondary infection.

Beak fractures are very common in captive cranes though fortunately the majority are minor. Especially during cold winter weather when probing behavior is impeded by frozen soil, fractures of the tip (1-4 cm) of either the upper or lower beak are seen. Trimming of over-long beak tips and providing safe substrates for probing help decrease this problem. More severe fractures also occur with the most common site being immediately in front of the nares on the upper beak. It is unclear why breaks at this site are common. Initial treatment includes control of hemorrhage and therapy for shock. Options for surgical repair are described under Common Surgical Procedures, this chapter. Regrowth of the upper beak will be minimal when the fracture is more than 3-4 cm from the tip. However, fractures of the lower beak ≤8 cm (less in small-billed species) from the tip can regrow. Providing visual barriers between cranes and human activity and between crane pairs will decrease the behavior at fence lines that puts cranes at risk for beak damage.

Ruptured Air Sacs

Ruptured air sacs are infrequently seen in cranes. Adult cases are generally minor; chick cases can be more serious. In most cases, handling trauma was the suspected cause. Some cases resolved without medical intervention; some involved extensive subcutaneous emphysema on the thorax, neck, and head (Fig. 8.4). For these, air was withdrawn using a syringe and needle several times from several different locations over the body, but each time the condition would return within 24 hours. Finally, latex drains (6-13 mm diameter tubes) or setons (narrow gauze loops) were surgically inserted through the skin and into the air spaces. These birds were given antibiotics and the drains or setons were cleaned with a 1% povidone iodine solution twice daily. Within 10 days, the birds returned to normal and the drains were removed.

Osteomyelitis

Bone infections can occur secondarily in open fractures contaminated before or during surgical procedures or in pododermatitis (bumblefoot). In the early stages, osteomyelitis is not evident on radiographs, but rather shows up some days later. Usually the first evidence is a hazy appearance of the bone

structure with some roughening of the trabecular outline. As the condition progresses, evidence of bone absorption and reactionary bone formation appear simultaneously. The periosteum may be elevated with some new bone formation occurring underneath (Fig. 8.11). Osteolysis may occur, especially if the infection is in conjunction with a foreign body such as a bone screw or intermedullary pin. Signs of possible spreading of the infection include widening or increase in size of osteolytic area, active periosteal response, loss of joint space, or soft tissue swelling. **Avian tuberculosis** is known to occur in several of the wild populations of North American cranes, and any bird from these flocks showing limb problems should be radiographed to search for bone changes similar to osteomyelitis or neoplasia.

Treat osteomyelitis with antibiotics or antifungals chosen by culture and sensitivity testing (Table 8.1) and with surgical debridement (removal of necrotic tissue). Radiographically, response to treatment is indicated by cessation of osteolysis and gradual redevelopment of normal bone structure (Douglas and Williamson 1970:44). The most common complications are delayed union of fracture sites or bone sequestra. Sequestra are seen as areas of bone separated from other nearby bone by a radiolucent (non-bony) zone. Sequestra are necrotic pieces of bone and are best demonstrated by lack of any change in appearance on serial radiographs.

Arthritis, reportedly uncommon in birds (McMillan 1988), when seen in cranes is secondary to trauma, infection, developmental limb problems, or articular gout. Iatrogenic arthritis is one sequel to carpal tenotomy often used to limit flight capability in crane colonies.

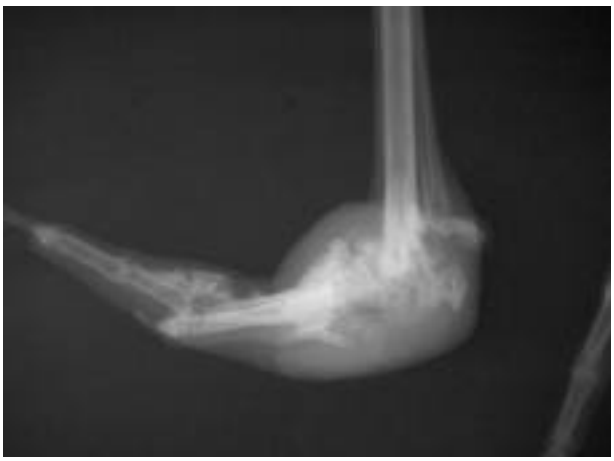

FIG. 8.11. *Osteomyelitis (radiograph).* PHOTO GLENN H. OLSEN

Egg Binding

Egg retention or egg binding is a rare condition usually associated with an abnormally formed egg (soft-shelled, abnormally large, or abnormally small), or a bird that has an abnormal pelvis or cloaca, is in poor condition, is hypothermic, or has low blood calcium. Occasionally an infection can lead to a retained egg. Clinical signs of egg binding include straining, lethargy, or depression. The egg is usually readily palpable in the caudal abdomen.

Initial therapy consists of providing a warm, quiet environment for the bird and lubricating the cloaca with surgical lubricant (see Appendix) or petroleum jelly. Medications should include fluid therapy, oxytocin, and intramuscular calcium (Calphosan, see Appendix) (see Table 8.1 for dosages and routes of administration). Occasionally, sedation is useful to relieve oviduct spasms. Antibiotic therapy should be considered in all cases where infection or contamination of the oviduct is suspected.

If the egg can be gently manipulated from the oviduct, this should be done. Breaking an egg is not recommended as the sharp fragments can lacerate the oviduct. If the egg does break, as many pieces as possible should be removed through the cloaca. If the egg is retained high in the abdomen (i.e., palpable for two days without movement), an exploratory laparotomy with surgical removal of the egg may be necessary.

Disorders of the Cloaca

Straining from egg retention, constipation, vent irritation, or diarrhea occasionally results in a prolapsed cloaca, oviduct, or part of the large intestine. Cloacal prolapses are seen in chicks with and without diarrhea. In some cases, stress is considered a factor. If possible, determine which organ is prolapsed and treat the cause before replacing the cloaca. Be sure to differentiate a prolapse from a protruding growth in the cloaca.

With a mild prolapse, it may be possible to replace the prolapsed tissues after lubricating them with surgical jelly. More severe cases may require bathing the tissues with a hypertonic solution (such as 50% dextrose) to reduce swelling prior to replacement. A gloved finger or smooth tube (syringe without needle) may help to reduce the prolapse. A purse-string suture may be required for several days to keep the tissues from prolapsing again. Once the purse-string suture is

in place, the crane must be watched carefully to make sure it can defecate normally. In laying females, a purse-string suture in the cloaca should be left in place only until about one day prior to oviposition. The underlying cause of the prolapse should also be treated. In severe cases, it may be necessary to remove necrotic tissue or amputate part of the prolapsed organ. Prognosis is guarded in these cases.

In some species, a tendency to prolapse appears to be inherited (Macwhirter 1987) although this has not been documented in cranes. Other cloacal abnormalities seen in cranes include a papilloma-like growth in a Sarus Crane and seasonal inflammation and vent soiling in laying females, for which no causative agent has been found.

Neoplasia

Several types of neoplasia have been reported in cranes including renal adenocarcinomas (Montali 1977; Decker and Hruska 1978), renal carcinoma (Montali 1977), lymphocytic leukemia (Montali 1977), granulocytic leukemia (Wei et al. 1986), and metastatic cholangiocarcinoma (Allen et al. 1985). A hematopoietic stem cell neoplasm occurred in one Florida Sandhill Crane at Patuxent. There is a higher incidence of adenocarcinomas in wild Mississippi Sandhill Cranes than in captive birds, but the cause of this situation is still under investigation. Chondroma-like lesions have been seen in wild Florida Sandhill Cranes and one Whooping Crane (M. A. Spalding, University of Florida, Gainesville, Florida, personal communication). Generally the incidence of neoplasia in both captive and wild cranes appears to be low.

Toxicology

Only a **few cases** of crane morbidity or mortality due to **specific toxins** have been reported. A likely assumption is that most substances that are toxic to birds in general are also toxic to cranes. Cranes are considered to be low on the food chain, and are therefore not as likely to be affected by toxic compounds through biomagnification (Mullins et al. 1978). However, cranes are relatively long-lived birds and therefore have the opportunity to slowly accumulate significant amounts of persistent chemicals.

Examination of two Whooping Crane carcasses and one embryo for DDT, DDD, DDE, and dieldrin demonstrated very low levels in all tissues (Lamont and Reichel 1970). **Pesticide levels** in fat from Sandhill Cranes from Florida and Nebraska were low (Lewis 1974). Samples from the same study in Texas contained high levels of heptachlor epoxide and smaller amounts of DDT, DDE, and dieldrin. Oklahoma samples one year had elevated DDT, DDE, dieldrin, and heptachlor epoxide levels. However, the next year, samples from the same Oklahoma site showed only low levels of DDE, with other pesticides not detected. Mullins et al. (1978) found low levels of pesticides DDT, DDD, DDE and dieldrin, and low levels of heavy metals, lead and mercury, in Greater Sandhill Cranes and eggs from Oregon and Idaho. Mercury levels were significantly higher in breeding age birds compared to eggs and young, indicating possible accumulation with age.

In 1989 and 1990, 58 Sandhill Cranes collected in Nebraska (as powerline mortalities) were analyzed for organophosphate and carbamate compounds and inorganics (Fannin 1992). Heptachlor epoxide, oxychlordane, DDE, and hexachlorobenzene were found in liver tissues. From opportunistic sampling of Whooping Crane carcasses and eggs since the 1960's, Lewis et al. (1992) reported that while DDT and mercury levels have declined following banning use of these substances as pesticides and fungicides, other compounds such as chlorinated hydrocarbons persist at low levels. Trace elements including aluminum, arsenic, cadmium, chromium, copper, selenium, and zinc were found at levels high enough to justify further monitoring.

Famphur, an organophosphate (o,o-dimethyl o-<p-(dimethylsulfamoyal) phenyl>phosphorothioate), was found at 69 ppm in the digestive tracts of two dead Sandhill Cranes in Gilmar County, Georgia (White et al. 1989). The brains from these two birds showed a 75% reduction in cholinesterase activity attributed to the effect of famphur. Organophosphorus compounds inhibit cholinesterase in the nervous system, disrupting synaptic transmission of nerve impulses. Death is usually the result of asphyxiation associated with failure of the brain respiratory center. Organophosphates and carbamates are known to be extremely toxic to wildlife, especially birds (Zinkal et al. 1978; Hill and Fleming 1982; Henny et al. 1985).

Both wild and captive Sandhill Cranes have died from **lead poisoning** when accidentally exposed. Mullins et al. (1978) found lead in samples of Greater Sandhill Cranes and eggs from Oregon and Idaho. Occasionally individual cranes, especially juveniles, have comparatively high levels (Franson and Hereford

1994). The source of lead has varied, but includes lead-based paints (Kennedy et al. 1977) and lead fishing weights and bullets (Windingstad et al. 1984). One wild Whooping Crane died after ingesting a small plastic encased battery or fish sinker (Snyder et al. 1992).

Zinc toxicosis has been seen in captive Whooping and Red-crowned Cranes after ingestion of zinc-containing metal objects, principally wire clippings from fence construction and zinc alloy coins. The clinical signs include depression, weakness, and lethargy. Recovery was rapid after surgical removal of the metal.

Mycotoxins produced by *Fusarium* sp. molds have caused death in wild and captive cranes. Between 1982 and 1987, an estimated 9,500 Sandhill Cranes in Texas and New Mexico died from an unspecified mycotoxin found on unharvested peanuts (*Arachis hypogaea*) (Windingstad et al. 1989). Cranes were observed standing or flying but unable to hold their necks straight or erect. Lesions observed at necropsy included multiple muscle hemorrhages and submandibular edema. Peanut-associated mycotoxicosis has also been seen in Demoiselle and Eurasian Cranes in India (ICF unpublished data). In 1987, deoxynivalenol toxicity, a grain-based mycotoxin present in pelleted feed, resulted in 4% (15 cranes) mortality and 80% morbidity at Patuxent (Olsen et al. 1995). Bioassays using quail or less valuable cranes are now in use with larger crane flocks to help detect unsuitable commercial feeds before they are fed. Carefully monitor food during long periods of warm, rainy weather.

Botulism, a paralytic disease caused by ingestion of *Clostridium botulinum* toxin, has killed cranes in at least one North American zoo. The toxin is produced anaerobically with the typical source being the decay of submerged carcasses of small animals. This kind of poisoning should be considered whenever cranes are housed in naturalistic water exhibits. Pay careful attention to water quality and the health of waterfowl using the exhibit. A commercial *Clostridium botulinum* Type C bacterin-toxoid has been used to protect cranes (Cambre and Kenny 1993).

Capture Myopathy

Capture (or exertional) myopathy, reported several times in cranes (Brannian et al. 1981; Hartman 1983; Windingstad et al. 1983; Carpenter et al. 1991), has been associated with trapping and restraint. It has also been seen in captive cranes after serious, traumatic injuries or after prolonged restraint in a sling. Predisposing factors described for other species (Boever 1986) and most likely applicable to cranes include fear, anxiety, overexertion, repeated handling, transportation of an exhausted animal, prolonged transportation, constant muscle tension, and restraining the bird with muscles cramped in unusual positions or for a prolonged time. In some species of mammals, metabolic acidosis may play an important role in capture myopathy (Harthoorn and Young 1974).

Clinical signs can range from peracute death due to cardiac failure, to painful, stiff movement, and swollen, hard muscles that are warm to the touch, and secondarily, trauma to limbs as the animal struggles. If the bird has survived for some days, there will be a reduction or loss of subcutaneous and abdominal fat. Serum chemistry levels, particularly creatinine kinase, lactic dehydrogenase, and aspartate aminotransferase, are often highly elevated and are useful in assessing the severity of changes in muscle tissue (Harthoorn and Young 1974; Brannian et al. 1981). Elevation in uric acid values associated with renal failure can result from increased lactic acid production, myoglobinuria, or impaired mobility and subsequent dehydration.

Gross lesions consist of numerous pale, streaked areas in the skeletal muscle and heart. Renal lesions, such as urate nephropathy, are also common. Pale mottled kidneys are described in one case involving a Greater Sandhill Crane (Windingstad et al. 1983), and urate deposits were described on the kidneys of an East African Crowned Crane (Brannian et al. 1981). Microscopically, lesions are characterized by extensive areas of myocardial and skeletal muscle degeneration and necrosis, and secondary inflammation.

Prevent capture myopathy by minimizing handling, by properly handling and transporting cranes (see Chapter 2), and by maintaining adequate levels of vitamin E and selenium in the diet. Treatment of the condition is supportive and consists of intravenous fluids, corticosteroids, antibiotics, vitamin E, selenium, and good nursing care. It may be necessary to keep the bird in a sling. Physical therapy is also important for recovery. If blood pH levels are below normal, the crane should be treated with intravenous sodium bicarbonate. If blood pH levels are unavailable, but acidosis is suspected, sodium bicarbonate may be administered IV at the rate of 4-6 mEq/kg body weight.

Orthopedics

Leg, Foot, and Toe Disorders

Fractures, and less frequently **dislocations**, are common in cranes. Fractures are usually associated with trauma. Pathological fractures associated with nutritional imbalances are not generally seen in captive cranes fed a formulated pelleted diet. Likewise, the occurrence of secondary nutritional fractures in wild cranes has not been documented. Fractures should be evaluated for location, articular involvement, bone density, periosteal response, and soft tissue involvement (McMillan 1988, 1994). Generally, midshaft long-bone fractures have better prognosis than fractures close to the bone ends or involving articular surfaces. Most long-bone fractures of cranes require surgical fixation (Fig. 8.12). Some fractures of pneumatic bones result in subcutaneous emphysema in and around the fracture site. Tendon and ligament injuries around the hock and foot are also seen. These are difficult to diagnose and require weeks or months to heal.

The **healing of crane bones** is similar to the process seen in other birds and mammals (Bush et. al. 1976). Healing in uncomplicated cases occurs in 3-8 weeks with pneumatic bones generally healing slower than medullary bones. Osteoporosis associated with disuse is a possible complication. Often some form of physical therapy is required for a crane to begin using a limb after a fracture has healed.

Leg injuries are common in cranes (Carpenter 1986; Curro et al. 1992; Olsen 1994), and include long bone fractures, stifle and tibial luxations, fractures and luxations of toes, spraddle legs, lateral rotation of the tibiotarsus, crooked or curled toes, and perosis. In chicks, rapid weight gain has been implicated, as have hatching problems and trauma. Exercise is a large factor in leg development. Crane chicks reared by parent cranes suffer much fewer leg abnormalities than do crane chicks raised by hand (see discussion of crane chick leg disorders in Chapter 5). Diets containing low levels of methionine or sulfur amino acids help reduce rapid growth and associated leg abnormalities in crane chicks (Serafin 1980, 1982). Correction of **crooked or curled toes** has been accomplished with splints (see Figs. 5.18 and 5.19).

Progressive osteoarthritis, especially in the hock joint of the legs, is seen in older cranes often after a long history of recurring mild lameness. Mineralization of tendon sheaths around the hock is a common radiographic finding. Medical therapy includes non-steroid anti-inflammatory drugs such as phenylbutazone and piroxicam to decrease the pain. Intra-articular and intramuscular injections of polysulfated glycosaminoglycans (Adequan) have been found ineffective in severe cases.

Bumblefoot (Pododermatitis)

Crane bumblefoot (pododermatitis) is an inflammation of the foot often associated with a bacterial infection (Fig. 8.13). Bumblefoot may start as pressure necrosis of the footpad, in many cases due to unequal weight bearing on the foot due to a lameness of the other leg. Generally, bacteria enter the foot by two routes, the first being an acute wound or laceration of the integument of the foot; the second route is

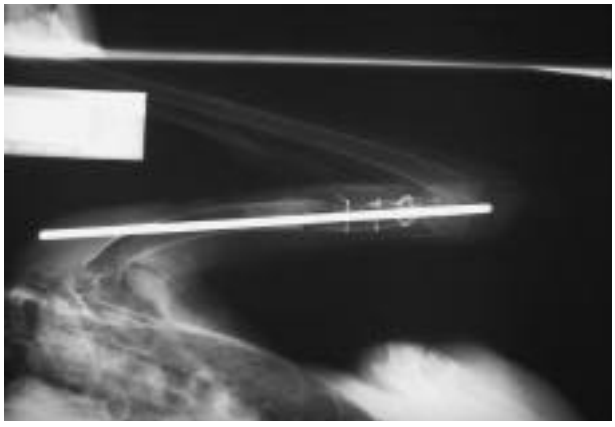

Fig. 8.12. *Long-bone fracture (radiograph) showing internal fixation.* Photo Glenn H. Olsen

Fig. 8.13. *Bumblefoot, Class IV–V.* Photo Glenn H. Olsen

through the cracking or sloughing of dead (devitalized) skin or scales that expose the underlying tissues. The disease is progressive. Infection can spread into the tendons and bones of the foot and can be debilitating. Rarely, infections spread elsewhere in the body.

A classification system developed for use in raptors (Remple 1993) is also applicable to crane bumblefoot. Five classes are used to describe the progressive course of the disease and to grade prognosis. Class I represents the early inflammatory response to a lesion and is characterized by tissue bruising or callus (hyperkeratotic reaction) with an excellent prognosis for recovery. In Class II, there is infection in the underlying tissues, localized loss of blood supply and death of tissue (ischemia and necrosis). With modern developments in treatment, prognosis is now good. Class III is characterized by extensive infection and gross inflammatory swelling characterized by serous, fibrotic, or caseous tissue reaction; prognosis is good to guarded. Class IV represents lesions with infection and swelling of underlying tissues resulting in inflammation of the ligaments or tendons (tenosynovitis), joints (arthritis; Fig. 8.13), or bone (osteomyelitis; Fig. 8.11); prognosis is guarded to poor. In Class V, the lesions of Class IV have worsened resulting in deformity, crippling of the limb, and loss of normal function; prognosis is grave.

Treatment should start with correcting any management problem or concurrent leg injuries contributing to the bumblefoot. Abrasive surfaces, such as concrete, can lead to foot injury. Medical care includes ensuring good nutrition through adequate diet plus vitamin and mineral supplementation. Vitamin A injections are given (30,000 IU/kg body weight or 1.0 mL/kg of Aquasol A intramuscularly once weekly). Surgery under general anesthetic is recommended in severe cases. The goals of surgery are to debride (cut away) necrotic and infected tissues to reduce antigen load. Skin edges should be brought together to enable first intention healing to occur (Riddle 1980). Prior to surgery, the site is thoroughly scrubbed with a stiff brush. After incising the skin, a culture for bacterial and fungal identification is taken from the deep tissues. If surgical debridement is not done, a sample for culture can be collected by fine-needle aspiration. Remove fibrotic and infected exudate and irrigate the wound with chymotrypsin and a broad-spectrum penicillin product such as piperacillin (Riddle 1980). Close the wound with a 4-0 or 3-0 nylon, simple interrupted suture pattern or use vertical or horizontal mattress sutures to relieve tension.

After surgery, the foot is kept bandaged until the wound is healed. If the surgical wound is on the plantar (bottom) surface of the foot, the bandage should be designed to reduce pressure on the surgery site and surrounding inflamed tissues. In raptors, a preformed styrene cast or ball bandage is used to accomplish this (Remple 1993). Variations on these techniques, modified for the anatomy of the crane foot, have been used with varying success. Keeping the bandage and surgical site dry can be a problem with cranes, especially those housed outdoors. A top layer of waterproof tape helps. Antibiotic therapy should begin before surgery or, if not before, immediately after surgery. A broad-spectrum penicillin, such as piperacillin, is recommended, often in combination with an aminoglycoside such as amikacin (see Table 8.1 for dosages). Antibiotic therapy should be modified based on the culture and sensitivity results, and should continue for a minimum of 14-21 days post surgery. The crane should be kept on a soft surface such as grass, padded indoor-outdoor carpeting, or deep bedding.

Bandaging and Splinting

Bandaging or splinting is frequently used in the treatment of orthopedic problems. To minimize stress, a bandage or splint should be completed as quickly as possible and should be as unobtrusive and lightweight as possible but still be adequate to protect and stabilize the body part. If the crane becomes agitated during the bandaging/splinting process, use sedation or a general anesthetic.

To **avoid damaging feathers** , especially remiges and rectrices, use Vetwrap (see Appendix, but avoid red or other brightly colored Vetwrap) or other self-adhesive tapes that do not adhere to the feathers. Masking tape or autoclave tape can be used, but they tend not to hold well to feathers, especially when wet. Adhesive tape should only be used if the feathers are first wrapped with gauze. On the scaled portions of the leg, adhesive tape is acceptable. In some situations, a layer of waterproof tape over the bandage is helpful to keep the underlying bandage dry. Check bandages and splints frequently to detect swelling, irritation, slippage, or removal by the crane. Bandages in young, growing birds should be changed every 48 hours.

The **Figure-8 bandage** (Fig. 8.14) is useful for fractures of the radius, ulna, and hand, or to support the wing during recovery from soft tissue injuries or developmental abnormalities. This bandage can be used alone, or if the bone ends are severely malaligned, in conjunction with internal fixation. The immobilization provided by the Figure-8 bandage can result in a temporary or permanent stiffening of wing joints preventing normal flight. For release birds, surgical fixation techniques are recommended to increase the chance of normal flight. Humerus fractures usually require internal fixation, and a temporary Figure-8 bandage and body wrap to limit wing movement.

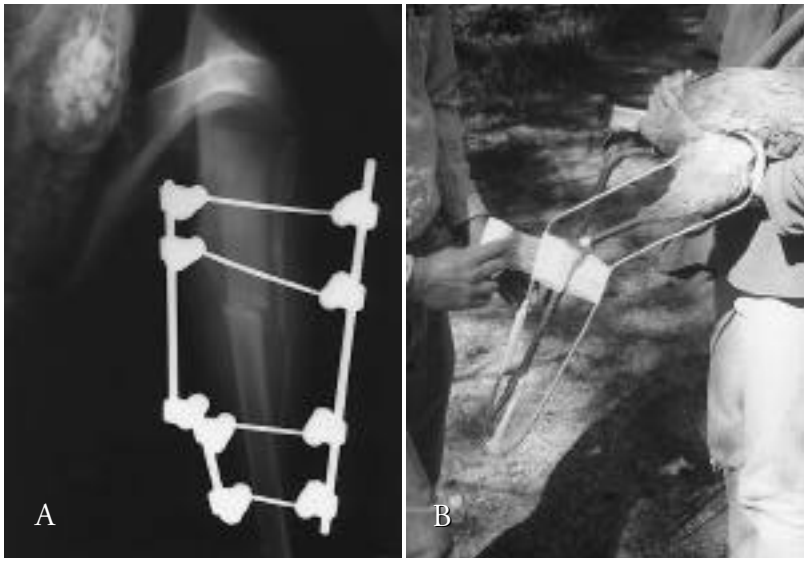

Fig. 8.15. *Two types of splints: A, Type II Kirschner Ehmer splintage for fracture of tibiotarsus in 41-day-old crane chick; B, Thomas splint (note wire makes a complete loop around thigh).* Photos Glenn H. Olsen (A) and David H. Ellis (B)

In cranes, broken legs often result in death. **Leg splints** may be useful in simple and open (compound) fractures of the tibiotarsus and tarsometatarsus (Olsen 1994). Leg splints are used alone or in conjunction with internal fixation. External fixators (Fig. 8.15a) have been used with both tarsometatarsal and tibiotarsal fractures with some success. Femur fractures, rare in cranes, generally require internal fixation (Howard 1990), although a Type I Kirschner Ehmer apparatus has also worked.

Some other leg splinting techniques (Figs. 8.15b and 8.16) are hinge splints and Schroeder-Thomas splints for both tibiotarsal and tarsometatarsal fractures. Spoon splints (usually with the spoon-end removed), fiberglass, and plaster splints are also used. Because the joints above and below a fracture site should be immobilized during the healing process, the crane should not be allowed to move about normally.

Leg prostheses have been successfully used in four cases of amputation below the hock for irreparable tarsometatarsal fractures. In one case, a custom-designed "human" orthotic was used; in the others, a section of PVC pipe was attached to the stump. Custom fitted wood and bamboo protheses have also been used.

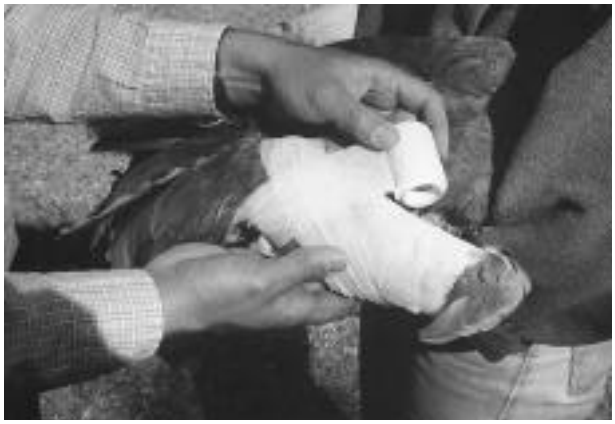

Fig. 8.14. *Figure-8 bandage.* Photo David H. Ellis

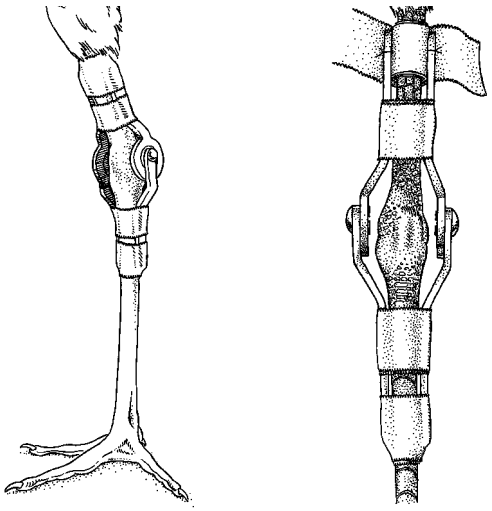

Fig. 8.16. *Hinge splint: The use of a hinge brace to provide medial-lateral support to joints with soft tissue injuries such as ligament tears has proven successful in the medical management of this type of injury in cranes (P. Klein, Humane Society, Washington, D.C., personal communication).* Art Kate Spencer

Because of crane anatomy, a flat or "shoe" type of splint is required to correct **phalangeal fractures** (Fig. 5.19), luxations, or to control knuckling during recovery from tendon or nerve injuries. Satisfactory **toe splints** are made of bental uminum rod and of reheatable plastic splint material (Hexcelite, Orthoplast, or Polyflex II). Often the whole foot is encased in the splint, with a spine of splint material extending up the tarsometatarsus to help stabilize the foot. Splints for curled digits in cranes chicks are discussed in Chapter 5.

When a serious leg or spinal injury occurs, supplemental support for the crane may be provided by a rigid framework with a **cloth sling** (Fig. 8.17). Some institutions use a cloth sling suspended by ropes from the ceiling. Cranes confined to slings often refuse to eat or drink, necessitating nutritional support by tube feeding. Some continually struggle with the constraining support system and require light sedation (midazolam, diazepam) to prevent self-injury. After removal of the splint or cast, another period of intensive management and therapy is required (Olsen 1994), and survival/success rates for cranes in slings are low.

FIG. 8.17. *Lorie Shaull attends a Sandhill Crane suspended in a sling.* PHOTO DAVID H. ELLIS

Anesthesia

Gas anesthesia, using isoflurane, is the best technique for sedation or surgical anesthesia in cranes (Ludders et al. 1989). Induction and recovery are rapid and smooth which is critical for the safety of the crane and handler. Halothane can also be used, but has been associated with a higher incidence of cardiac and respiratory problems. Although pre-anesthetics such as midazolam or tiletamine-zolezapam (Table 8.1) can be used, cranes are generally induced by mask and then intubated (2.5-6.0 uncuffed endotracheal tubes). Injectable anesthetics such as tiletamine-zolezapam or ketamine combined with diazepam, midazolam, or xylazine (Table 8.1) can be used, but respiratory and cardiac complication rates are higher and the crane must be monitored carefully and preferably held in a small padded room during recovery. Yohimbine has been used on several occasions to speed recovery when xylazine has been used. Local anesthesia in cranes is possible using small amounts of lidocaine (0.25-0.5 mL in adult cranes) or another local anesthetic.

Common Surgical Procedures

Laceration Repair

Lacerations are common in cranes. Cranes occasionally cut themselves (especially in the neck) with their sharp toenails (especially the inner nail) when they struggle during capture. This injury occurs 2-3 times each year among the 100+ crane chicks raised at Patuxent. Lacerations also occur from sharp objects in the pen and from aggressive pen-mates. Common sites for lacerations include the head, dorsal neck, carpal area, and the legs. Local or general anesthesia is used during repair of lacerations.

First, control hemorrhaging with compression on the wound site. For small wounds, ferric chloride hemostatic powder (Macwhirter 1987), ferric subsulfate, or Monsell's solution will stop bleeding. After the hemorrhage is controlled, gently pluck body feathers (not remiges or rectrices) around the wound site if needed, but avoid restarting the hemorrhage. Clean the wound with a dilute solution of antiseptic such as povidone iodine (1%), warm saline solution, or chlorhexidine. If the wound edges are not fresh, they should be debrided (cut back to fresh tissue). Suitable suture material can include 3-0 or 4-0 nylon or similar sized polyglycolic acid absorbable thread. Suture patterns generally reflect the nature and extent of the laceration with simple continuous, simple interrupted, and horizontal mattress patterns (Fig. 8.18) being the most common used in cranes. Cranes sometimes try to remove bandages with their bills, so reinforce accordingly. Non-absorbable sutures can be removed after 10 to 14 days; absorbable sutures are normally left in place.

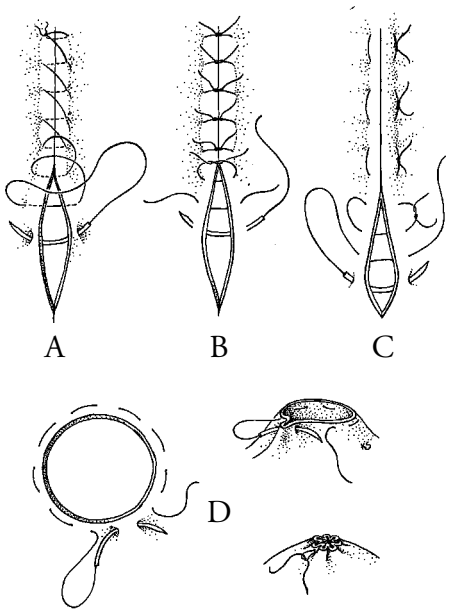

Fig. 8.18. *Sutures: A, simple continuous; B, simple interrupted; C, horizontal mattress; D, purse string.* Art Kate Spencer

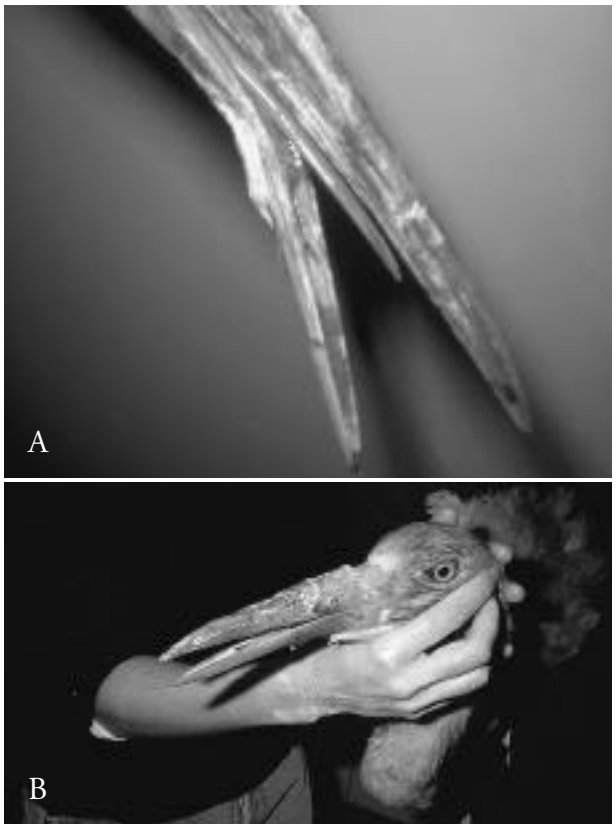

Fig. 8.19. *Crane beak repairs: A, fractured lower mandible before repair; B, fractured maxillary after repair with dental acrylic.* Photos Glenn H. Olsen

Repair of Fractured Beaks

Minor fractures of the tip of a beak can be managed without anesthesia in two ways. The beak can be trimmed at the point of fracture. Trimming works well for fractures occurring on the distal 2-3 cm of the beak, but may result in profuse bleeding. Alternately, the fracture can be stabilized with cyanoacrylate glue (surgical glue, "super glue") while the blood vessels constrict. After 2-5 days, the fracture fragment is trimmed off. Cyanoacrylate glue or dental acrylics can also be used to provide homeostasis and protect the stump of a fractured beak. If only the upper or lower beak is fractured, the protruding section of the unfractured beak is gradually trimmed back using a hand-held grinding tool (Dremel tool; see Appendix) or surgically removed using radio-cautery for hemostasis (Ellman radio surgery unit; see Appendix).

More serious fractures require surgical management under general anesthesia. One method of repair uses self-curing dental acrylics or hoof acrylics (Fagan 1982; Altman 1984; Frye 1984; Wolf 1985; see Appendix). The fracture site is cleaned, and any open wound is treated. The fracture is then manually aligned, and the beak covered with a layer of acrylic (Fig. 8.19). Because acrylics generate heat when curing, we sometimes surround the acrylic with cold packs to minimize damage to soft tissues. Unless the fracture site is very stable, the acrylic material will require reinforcing with Kirschner wires or stainless steel plates. These are applied longitudinally along the sides, top, or bottom of the beak, over the first layer of dental acrylic, and held in place with another layer of acrylic. For added stability, one or more Kirschner wires are implanted at right angles through the acrylic splint and beak. The final step in the process is to remove sharp edges by smoothing and shaping the outside surface of the acrylic with a high-speed, hand-held grinder (Dremel tool). Acrylic splints are generally left on the beak for 4-6 weeks.

Other techniques that have been used for beak repair include bone plating, Kirschner-Ehmer-type external fixation, and intramedullary pinning (Howard 1989). Frequently, cranes will not eat for the first day or two after surgical repair. Feeding through a pharyngostomy or esophagostomy tube may be less stressful for these patients than repeated oral tube feeding and handling of the beak. Rates of successful repair with severely displaced fractures have been low, especially in species such as the Siberian Crane which does not stop using its beak for probing after surgery.

In cases where the fractured beak fragment has been lost, beak prostheses have been designed using moldable acrylics and attached with intramedullary pins, wire, or acrylics (Greenwell et al. 1989). In cranes, a beak prosthesis generally needs to be replaced every 3-6 months due to the wear associated with the crane's probing activities. For these reasons, a beak prosthesis is probably more appropriate for an exhibit crane, not a breeding crane. Several captive cranes have fed normally and have survived for years in captivity with beaks shortened unilaterally or bilaterally by as much as half the length of the normal beak (Howard 1989).

Endoscopic Examinations

The first avian endoscopic examinations (using rod-lens systems) were in the early 1980's (McDonald 1982) for determining sex in monomorphic psittacines. The technique is useful in cranes both for determining sex and for diagnosing a variety of abdominal and respiratory disorders. A rigid endoscope (1.9-2.7 mm outside diameter, 30° view, 17-19 cm length; see Appendix for source) is most useful. A 150-watt light source is attached via a flexible fiberoptic cord. A system using a handle-mounted, battery pack (ophthalmoscope/otoscope handle) with a focusing ocular piece on a rigid tube (Medical Diagnostic Systems, see Appendix) has proven useful in some field situations and is about one sixth the price of the rod-lens endoscopes. The disadvantages of this more portable system are reduced light transmission and poorer optics. Flexible endoscopes have also been used for gastrointestinal and tracheal examinations and for foreign body retrieval from the upper gastrointestinal tract and trachea (Howard et al. 1991). A human bronchoscope with extra channels (i.e., for insertion of other instruments) is recommended. We have used such units coupled with laser cautery to remove tumors from the accessible portions of the gastrointestinal tract and the trachea.

The **endoscope is sterilized** before use. One method is to expose it to ethylene oxide gas. After exposure to the gas, the instrument needs to be aired for 8-12 hours before use. Ethylene oxide also poses a human health hazard and manufacturer's safety recommendations should be followed. A second, safer and easier, method is to soak the endoscope for 15-20 min in 2% glutaraldehyde (Glutarex, see Appendix). Soaking for more than 2 hours may damage optics. After soaking, the endoscope is rinsed with sterile, distilled water prior to use. Two separate rinses of 3-5 min each are recommended. Basic endoscopy is described in Taylor (1994).

The site is prepared for the surgical procedure after the patient is anesthetized (isoflurane and ketamine have been successfully used for endoscopic examinations). We strongly advise against performing endoscopic examinations on physically restrained, but non-anesthetized cranes.

There are at least six commonly used **endoscopic entry sites** on each side. Choice depends on the organ system to be view. The sites are: (1) the ventral midline or just lateral to the midline at the posterior margin of the sternum for examination and biopsy of the liver, (2) one side of the ventral midline near the pelvis for examination of the gastrointestinal and urogenital tracts, (3) lateral to the cloaca also for the gastrointestinal and urogenital tracts, (4) the flank behind the last rib for the genital organs and posterior lungs, (5) the flank in front of the last rib also for the genital organs and posterior lungs, and (6) dorsally between the second- and third-to-last ribs, slightly below the vertebral column (and just anterior to the leg) for examination of the lungs, genital organs, and heart. In Siberian Cranes, hemorrhage has frequently occurred when the entry site is behind the last rib. Therefore, an entry site between the last two ribs is preferred for this species. Rigid and flexible endoscopes have also been used to visualize lesions in the trachea, esophagus, and cloaca.

Contraindications for endoscopic examination include severe disease conditions that would prevent general anesthesia, obesity, fluid in the abdomen (ascites), and the presence of a large developing egg. Possible complications include trauma to organs; laceration of a blood vessel, liver, or spleen resulting in serious hemorrhage and occasionally death; subcutaneous emphysema from air leaking from an air sac through the hole made in the body wall; and the possibility of sepsis at the surgery site. The risk of subcutaneous emphysema can be lessened by placing an absorbable suture in the body wall and another in the skin upon exiting an endoscopic site, or by using tissue glue (cyanoacrylic glue) to seal the surgery site. Trauma to organs, hemorrhage, and sepsis are all controlled best by using proper techniques. We recommend prior training to avoid mistakes and to enable proper interpretation of tissues viewed through the endoscope.

Ventriculotomy/Foreign Body Removal

Cranes frequently pick up bright or novel objects and occasionally swallow them. These objects frequently lodge in the ventriculus. Clinical signs reported in Sarus Cranes include lethargy, labored standing, hock sitting, and diarrhea (Bush and Kennedy 1978). Gastrointestinal bleeding and signs of heavy metal toxicosis (lead, zinc, etc.; see Poisoning) have been seen. One Whooping Crane in Maryland died from ingesting a nail that punctured the ventriculus. In another case, a Florida Sandhill Crane that ingested a gold earring suffered severe clinical signs. Two Whooping Cranes that swallowed wire survived. In one case, the wire penetrated the gizzard and became walled off in a necrotic mass in the abdomen. In the second case, the wire penetrated the gizzard mucosa and was found in the gizzard muscle layers. Operations to remove the wire were successful in both cases.

If clinical signs appear, take **radiographs** to confirm the presence of a metallic foreign body (Fig. 8.20). Unfortunately, not all foreign bodies are evident on radiographs. **Surgery** can be performed to remove the object (if it is likely to poison the crane or pierce the gastrointestinal tract). Surgery (a ventriculotomy or proventriculotomy) is performed under general anesthesia using isoflurane. If possible, fast the crane 12-24 hours to reduce gut contents. Place the crane in right lateral recumbency and incise the skin and muscle parallel to the last rib. The ventriculus is exteriorized, packed off, and incised through the *musculus intermedin* at the posterior end of the ventriculus (or through the less muscular proventriculus). The foreign object is removed and then the ventriculus is closed with 4-0 absorbable suture material in a 3-layer closure. First, close the mucosa/submucosa with simple interrupted sutures. Second, close the ventriculus muscle wall with both horizontal and vertical mattress sutures. Third, close the adventia with interlocking continuous sutures. The abdominal muscle wall and skin are closed with 4-0 absorbable sutures. Reduce food intake for 5 days post surgery and treat the crane with antibiotics (Table 8.1). Feces will return to normal within 10 days after surgery (Bush and Kennedy 1978). Only recently has endoscopy proven useful for retrieval of stomach foreign bodies in adult cranes.

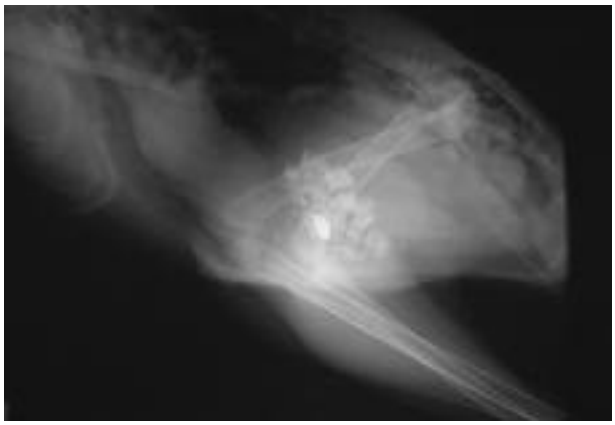

Fig. 8.20. *Ingested object (radiograph).* Photo Glenn H. Olsen

Preventive Medicine

Preventive medicine should include annual health checks. Each bird needs a physical exam, a blood count and blood chemistry profile, a screening for likely or common infections (e.g., Salmonella, EEE, IBDC, TB), and a fecal parasite analysis. For cranes entering or leaving the colony, impose a 30-60 day quarantine with disease screening. Prophylactic treatment (for parasites, etc.) and vaccination (EEE, botulism) schedules should be developed to meet the needs of the flock. The Whooping Crane Health Advisory Team has published detailed preventive medicine protocols (Langenberg and Dein 1992) that are useful for other species.

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